

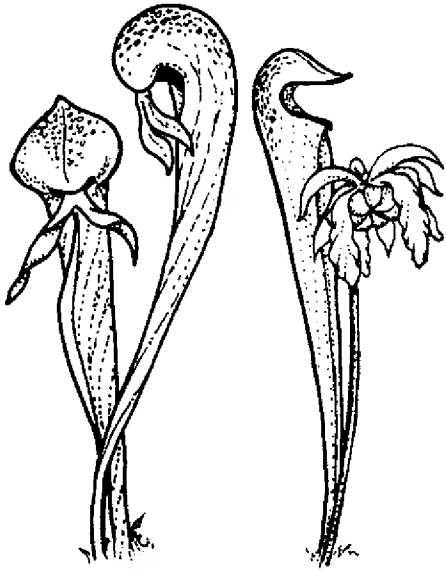
CARNIVOROUS PLANT NEWSLETTER

Journal of the International Carnivorous Plant Society

Volume 50, No. 3

September 2021





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Journal of the International
Carnivorous Plant Society
www.carnivorousplants.org

Volume 50, Number 3
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Front Cover: *Drosera falconeri* in full growth in the Darwin area, Northern Territory, Australia. Photo by R. Nunn. Articles on page 118 and 133.

Back Cover: *Drosera fulva* at the end of the wet season, growing at Noonamah, Northern Territory, Australia. Photo by R. Nunn. Article on page 118.

Carnivorous Plant Newsletter is dedicated to spreading knowledge and news related to carnivorous plants. Reader contributions are essential for this mission to be successful. Do not hesitate to contact the editors with information about your plants, conservation projects, field trips, or noteworthy events. Advertisers should contact the editors. Views expressed in this publication are those of the authors, not the editorial staff.

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Date of effective publication of the June 2021 issue of Carnivorous Plant Newsletter: May 11, 2021.

The ICPS is the International Cultivar Registration Authority (ICRA) for the names of cultivated carnivorous plants according to the International Code of Nomenclature for Cultivated Plants. Send relevant correspondence to the ICPS, Inc.

Carnivorous Plant Newsletter is published quarterly in March, June, September, and December by the ICPS, Inc., 2121 N. California Blvd., Suite 290, Walnut Creek, CA 94596, USA. Periodicals postage paid at Walnut Creek, CA and additional mailing offices. Postmaster: Send address changes to ICPS, Inc., 2121 N. California Blvd., Suite 290, Walnut Creek, CA 94596, USA. Printed by Allen Press, Inc., 810 E. 10th Street, Lawrence, KS 66044. Logo and masthead art: Paul Milauskas. © 2021 International Carnivorous Plant Society. All rights reserved. ISSN #0190-9215

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SURVIVING THE PANDEMIC

ALISON CARRINGTON-TARDIF • Sebastopol • California • USA • alison@californiacarnivores.com

This is a universally accepted truth: organisms which best adapt to suit their environment have the highest chances of survival. No plant, animal, or person exists in stasis, and the natural history of our world is rich with stories which all center a common theme: adapt or perish. As cultivators and sellers of carnivorous plants, the stakes in our nursery aren't usually so dramatic. When COVID-19 struck and brought the world to a halt, it was immediately clear we were facing a test unlike any other. What wasn't clear was if – and how – California Carnivores would survive the pandemic.

At the end of March 2020, the doors were closed to the public as Sonoma County entered a strict Shelter in Place ordinance. Damon Collingsworth and Daniela Ribbecke needed to lay off staff. Damon and Daniela took on all the work that had previously required a team of 5 people. They packed and shipped mail order, kept on top of office work and customer service, made new plants, and maintained the collection all while having no idea when it would end or what would come next. As March dragged into April, they acknowledged that what we all were experiencing would be a marathon and not a sprint. What wasn't clear was how long they could maintain momentum and survive.

Every week of those first few months was more tiring than the last. So many loyal customers were wonderful and understanding of the reduced capacity and speed, but they also experienced a new level of anger directed in ways never previously experienced. Because of the public closure, Damon and Daniela had to turn people away at the gate; most were understanding, but some were frustrated and angry. For more than a month, they agonized over every decision: can we safely bring anyone back yet? Are we making the right choice keeping our staff at home? As a small business, we were so lucky not to lose sales during this time, but neither were we sure what we could continue to count on.

As luck would have it, a sort of houseplant Renaissance evolved as lockdowns across the world extended through the year. People who had never been interested in gardening looked around their spaces and quickly sought to fill them with life. As this collective interest in plants grew, so did sales; it seemed they quadrupled overnight. In June 2020 it felt safe with appropriate protocols to start slowly bringing team members back, one by one, to catch up on sales and begin planning once again for the future. I was hired to handle the administrative duties and customer service.

California Carnivores has always been many things to all of us – retail nursery, lush botanical wonderland, meeting place for horticulturalists and hobbyists of all ages. We've always known how lucky we are and how special it is to be able to devote our lives to something so unique and meaningful. But the realities of operating a highly specialized retail space in an increasingly uncertain world had been creeping in at the edges for a while, even before Covid struck. Online sales did not just carry us through the pandemic; they allowed us to thrive as a business in ways that weren't previously possible. We made the decision to become online-only and close our public retail space. This is an adaptation that was difficult emotionally but necessary to the continued success of the business.

As plant people, we delight in the natural world and have devoted our lives to preserving its many wonders. As businesspeople, we are immeasurably grateful that social media and our digital connections have allowed us to survive a global tragedy. Change is a critical component of every life cycle, and every organism must adapt to these changes in order to survive. We feel so much hope when we think about the millions of new plant people who now enjoy a deeper connection to nature thanks to the tools of social media, and we will continue to use these tools to share our love, knowledge, and passion for carnivorous plants with the world.

WORLD CARNIVOROUS PLANT DAY: A WORLD-WIDE SUCCESS

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For our first World Carnivorous Plant Day (WCPD) Kenny Coogan and Carson Trexler recruited a range of hobbyists, nurseries, professors, and authors. Everyone we contacted was very generous with their time. Jeremiah Harris' international connections were immensely helpful in finding content. John Kim edited all 20 videos, a huge task, that is much appreciated. Thank you Krzysztof Banaś for the wonderful event idea!

In the first 24 hours of the videos being released, there were 2272 views on our YouTube channel <https://www.youtube.com/user/icpstv> and there were 5294 views on our Facebook page <https://www.facebook.com/InternationalCarnivorousPlantSociety/>.

The videos are also available on our website at

<https://www.carnivorousplants.org/about/WCPD/WCPD2021>.

- Happy WCPD Video (featuring hobbyists, professors and nurseries from 12 different countries. Video also featured dozens of students showcasing their CP inspired artwork and their completed ICPS coloring sheets and mazes.
- Kazuki Tagawa: Japanese Sundews Catch Prey in Groups, & Other Facts
- Adam Cross: Questions about the Carnivorous Plants of Australia
- Alastair Robinson and Adam Cross: The Endangered World of Carnivorous Plants
- Bartosz Płachno: The Scientific Importance of Carnivorous Plants
- Ulrike Bauer: The Complexity of Nepenthes Adaptations
- Rob Cantley, Diana Cantley, and Professor Entwistle: A Tour of Borneo Exotics and their role in conservation
- Chris Thorogood: Oxford Botanic Garden
- Matze Maier: Green Jaws: Carnivorous Plant Care for Beginning Growers
- Greg Allan and Cindy Chiang: *Byblis*, an Introduction to the Rainbow Plants
- François Mey: Meeting Botanical Artist François Mey
- Author Joanne Mattern reads her book *Disgusting Plants*
- Author Mary Batten reads her book *Hungry Plants*
- Rachel Wilson, Leslie Bradbury, and Kenny Coogan: Carnivorous Plants as Educational Models
- Ulrike Müller: Hydrodynamics of Bladderwort Traps
- Tom Givnish: Discovering Carnivory in a Bromeliad, and the Modern Definition of CPs
- Damon Collingsworth: Growing Mexican Butterworts
- Maggie Chen: Giant Venus Flytraps and How to Grow Them
- Barry Rice: Conservation and Cultivation of Carnivorous Plants
- Annual Photo Contest

WCPD 2021 merchandise is available at: <https://world-carnivorous-plant-day.creator-spring.com>

Options include: various types of clothing, face masks, mugs, and tote bags with the WCPD 2021 logo. A proportion of the proceeds will support ICPS conservation and education initiatives. Shipping is worldwide.

Save the date for next year! May 4, 2022. If you are interested in participating as a contributing speaker for World Carnivorous Plant Day in 2022 contact either Carson or Kenny. If you have a student or child between the ages of 12 and 16 who would like to interview an expert from another country, please contact Kenny.

WORLD CARNIVOROUS PLANT DAY PHOTO CONTEST

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We concluded this year's inaugural World Carnivorous Plant Day with our annual photo contest. Our judges were:

- Mindy Lighthipe draws and paints the beauty of nature. She believes in protecting the earth and does so by educating people through her art. Her goal is to record the earth's species in the 21st century and educate people to preserve it. <https://www.MindyLighthipe.com>
- Bertha Vazquez is the director of the Teacher Institute for Evolutionary Science, a non-profit foundation passionate about helping teachers explain evolution in an effective and engaging way. <https://www.tieseducation.org>



We had 15 countries represented and around 150 entries! A video showing all entries can be found at: <https://www.carnivorousplants.org/about/PhotoContests/2021PhotoContest>

Thank you to everyone who participated. Start snapping photos now for next year's contest. Winners received a 1-year membership to ICPS.



Category: People and Plants (Wacky or serious photo of carnivorous plants and the people who love them).

Phillip Bernie from Australia. *Sarracenia* sp. with a spectator.



Category: Portraits of Carnivorous Plants (a single plant or collection).
Stephane Joly from France. *Drosophyllum lusitanicum* and prey.



Category: Carnivorous Plants in the Wild (photos taken of plants in their natural habitat).
Krzysztof Banaś from Poland. *Drosera intermedia*.

FREQUENCY OF TRAP CLOSURE AND CAPTURE OF PREY BY *DIONAEA MUSCIPULA* IN THE FIELD

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Abstract: Measurements in the field on dry days indicate that when the traps of *Dionaea muscipula* (Venus' flytraps) close, they capture prey about 40% of the time at one site and 44% at another. The 60% and 56% of traps that close but do not catch prey reach their maximum closure on average in about 10 minutes, begin reopening in about 7 hours and completely reopen in 22 hours. Traps that have captured prey tighten along their rims, sealing the lobes and forcing the margins outward into a "narrowed" condition that takes 4 to 6 hours to achieve. Reopening of narrowed traps begins, on average, after 9.2 days and they are fully open in 12.8 days. A few traps that capture large prey never reopen.

In dry weather closure frequency of traps capable of closure was low, averaging 3.5%/day at one site and 3.1%/day at another. At both sites the capture frequency of traps capable of closing was only 1.4%/day. The low frequency of capture, and high percentage of closures that result in capture, indicate that the low capture frequency is due to a low number potential prey entering and triggering the trap rather than a low efficiency of capturing the prey that enters.

Traps that close, but fail to capture prey, have been observed to be triggered by insects, spiders, snails, and even toads that were far too large to be captured. A few captured beetles have been observed to force their way out of trap lobes.

Dionaea traps capture prey at all hours of the day. The capture frequencies are variable and are not significantly higher in any specific time period. Both nocturnal and diurnal animals were captured.

Introduction

Dionaea muscipula has an elaborate snap trap that has attracted nearly universal attention. Numerous popular descriptions describe flies being attracted to copious nectar secreted from glands along the edge of the trap but with no evidence cited. Adrian Slack (1980) states "Many insect visitors are undoubtedly attracted to the trap by the scent of the nectar supply, which is probably further advertised by the red coloration of the lobes, but a large number are casual visitors which are not nectar feeders." These ideas probably started with very convincing videos from the BBC, which have been reviewed by Williams and Hartmeyer (2017). While the actual frequency of capture is not mentioned in these sources, the suggested effectiveness of the attractants infers that captures should be frequent.

Actually, the limited observations that have been made suggest that prey capture is relatively infrequent (Williams & Hartmeyer 2017; Lichtner & Williams 1977) and a visit to their native habitat in North Carolina on a sunny day can easily confirm that most of the traps are open although they are capable of rapidly snapping shut if stimulated. Williams (1980), describing earlier measurements he made with Frank Lichtner, says that on a day with no rain 2.0% of 201 traps closed without capturing prey and 3.0% of traps captured prey. At this frequency of capture a typical trap would catch prey about once every 50 days. This single measurement made in the plant's natural habitat,

and similar measurements made by Williams and Hartmeyer (2017) on greenhouse and garden plants should be supplemented by more observations of plants in the wild.

The low capture frequencies of *Dionaea* could be due either to a small number of visitors to the traps, to a small number of the visitors triggering the traps, to ineffectiveness of triggered traps in capturing prey, or to a combination of these factors.

We have observed trap closures and captures by *Dionaea* plants growing in a stand grown from seed broadcast on the gulf coastal savannah of Florida 15 to 17 years ago (Site 2) and a second smaller population that descends from about six *Dionaea* plants transplanted about 15 years ago (Site 3). These plants descend from seeds originally harvested in the Carolinas (Williams & Scholl 2021a).

Our object is to measure closure frequencies and capture frequencies of *Dionaea* traps as well as rates of reopening of traps in a wild area similar to their native habitat. We also observed actual captures in the field and the factors that cause closure of the traps that fail to capture prey.

Methods

Plants in two stands of *Dionaea muscipula* were used in this study. Both are described by Williams and Scholl (2021a). One is a wet site near a small stand of Titi trees (*Clitonia monophylla*) that have covered the ground with leaf litter. *Sphagnum* is growing among the *Dionaea* plants (Site 3). The second is the neighboring Florida population that originated from seed, descended from plants from the Carolinas broadcast 15 to 17 years ago (Site 2). These are growing among scattered tall pines in a grassy area typical of the coastal savannah.

Photography and videography were done using a Bushnell NatureView camera mounted horizontally on a TopView Quick-Mount camera stand, above the plants being observed. The camera recorded a 25 × 35 cm field of view as still photographs and a 15 × 24.5 cm field of view as video. Every 5 minutes a photograph was taken, immediately followed by 1-minute video. The photographs were overlain with a grid and sequentially placed in a slideshow, which was scanned to observe the times of trap closure. Videos recorded after trap closure were then observed to determine if the traps reopened or narrowed from the capture of prey. When possible the cause of trap closure was also observed. Details of the camera methods are described by Williams and Scholl (2021a).

Method of measuring frequency of trap closure was the same as used by Williams and Scholl (2021b).

Trap closure occurs in a fraction of a second and is the flipping of the trap from the open position to the position in which the bristles along the edge of the leaf cross each other and only small gaps exist along the rim of the trap.

Trap narrowing (Fig. 1) occurs within minutes of the capture of prey. Mechanical stimulation from the struggling prey and later chemical stimulation from decomposing prey causes trap narrowing (Williams & Pickard 1980). Narrowed trap's lobes close tightly together, sealing along a smooth area just inside the edge of each trap lobe. The bristles along the edge of a narrowed trap are forced outward giving a narrowed trap a distinctive look. Narrowed traps can be used as a way to identify closed traps that have captured prey.

Active traps, as used in this study, refers to traps that are capable of closing.

Frequency of trap closure per day = closures/active-traps/day.

Frequency of capture per day = narrowed-traps/active-traps/day.

Frequency of closure without capture = (closures/active-traps/day) - (narrowed-traps/active-traps/day).

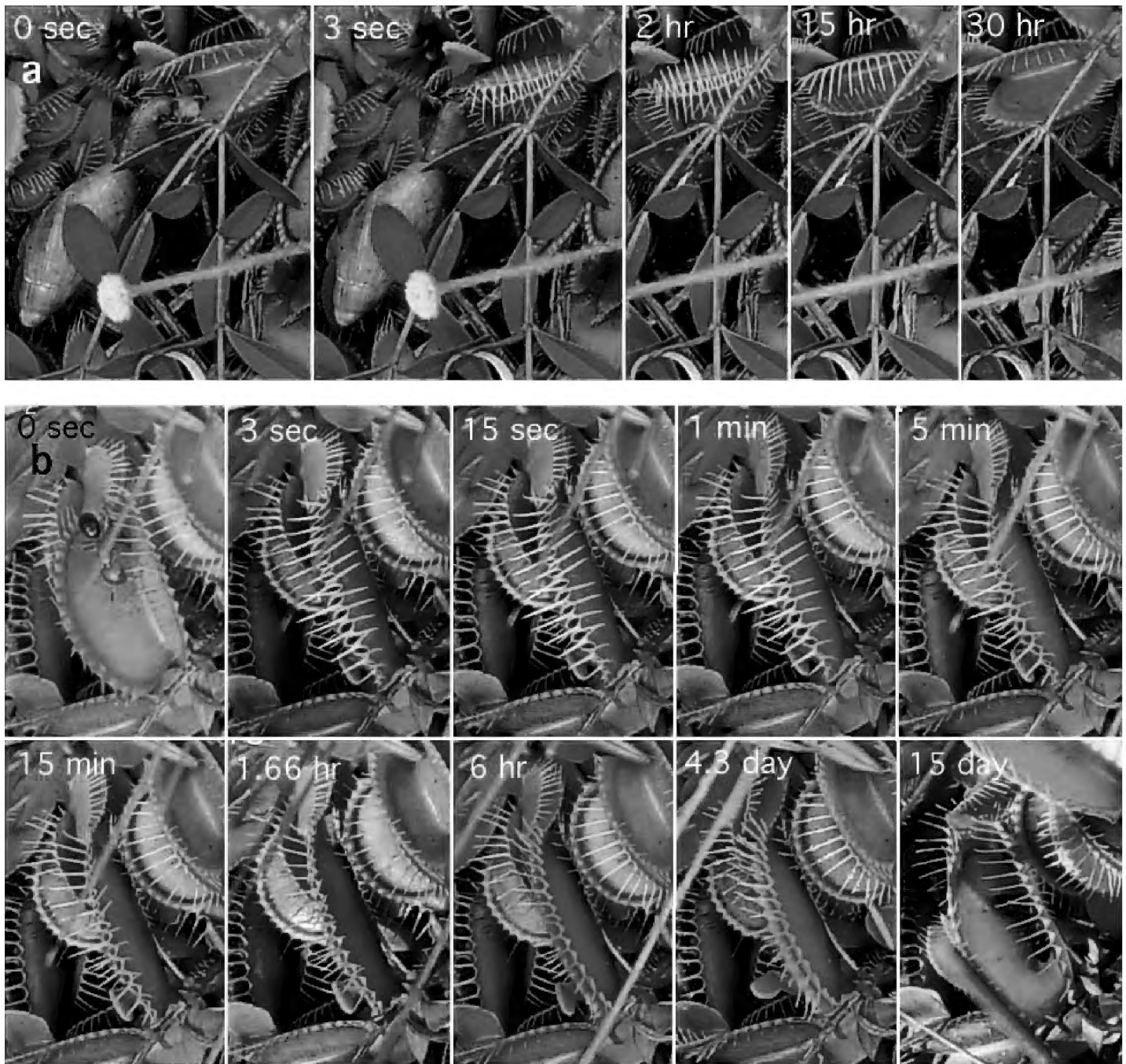


Figure 1: a. Closure without capture. A trap set off by a large snail. The trap is stimulated to close quickly, the snail pulls its head free and the trap opens after dark in 15 hours and fully opened by 30 hours. b. Capture and trap narrowing. A large ant triggers the trap to close. The ant's struggles stimulate the trap lobes to press together tightly sealing along their rims in about 5 minutes. They continue to press more tightly together for 6 hours causing their rims to flex outward in the "narrowed" position. After about two weeks the trap is fully reopened, revealing the undigested exoskeleton of the ant.

Measuring frequency of closure and capture at different times of day

Trap closures and captures were tallied in eight, three-hour periods: Early morning (03-06), Morning (06-09), Late morning (09-12), Early afternoon (12-15), Late afternoon (15-18), Evening (18-21), Night (21-00), Late night (00-03). Times are EST on a 24-hour clock.

Measuring reopening time

Reopening time of a trap was measured by subtracting the time when the trap was closed from the time when the trap was fully reopened. Both narrowed traps, that have captured prey, and closed traps, that have not captured prey, were measured in the same way. We also determined the time between trap closure and the time when the trap first began to move its lobes apart.

Measuring proportion of traps that are open and in other states

A 15 cm by 24 cm area was observed during dry weather on five dates between 7/17/20 and 10/10/20 and the proportion of traps in open, closed, narrowed, and reopening states was measured. The traps too young and too old to be active were disregarded but the traps which had captured prey and were in a narrowed state were included in “total traps” for this measurement. Traps in the reopening state can be reopening from the narrowed state or the closed state.

Results

Trap Narrowing

At least two stimuli to a trigger hair, or one stimulus to each of two hairs of a *Dionaea* trap within a period of 20 seconds will cause the lobes of the trap to come together, closing it (Brown & Sharp 1910; Fig. 1a). Additional stimuli will cause the lobes of the trap to tighten further and press against each other. When prey is captured, its struggles deliver numerous stimuli to the trap’s trigger hairs, each of which causes the lobes to press more tightly against each other, ultimately pressing so hard that the edge of the lobes are forced outward and the lobes are sealed tightly along a smooth region just inside their outer edges (Lichtner & Williams 1977; Fig. 1b). Traps with this appearance are called “narrowed” (Lloyd 1942) and form a digestive cavity, which has been called a “green stomach” (Scherzer *et al.* 2015).

Traps stimulated to close which have no further stimulation do not close tightly and reach their maximum closure in about 10 minutes. They begin reopening in 7 hours and are completely reopened in 22 hours.

Traps that have captured prey receive additional mechanical stimulation from the prey’s attempt to escape, and later by chemical stimulation during the prey’s decomposition (Lichtner & Williams 1977). This further stimulation causes the trap to tighten, sealing its lobes along a smooth strip along each lobe resulting the trap reaching a narrowed state. The initial trap closure typically takes less than a second but it takes traps 4 to 6 hours to become fully narrowed and days to weeks to reopen (Table 1).

The traps that failed to capture prey began to open in an average of 0.29 days and were fully open in an average of 0.90 days (Fig. 1a, Table 1). Five traps that have captured prey and narrowed began to reopen in an average of 9.2 days and were fully open in 12.8 days (Fig. 2a, Table 1), but when large prey is captured, or when the trap is older, opening can take much longer and may not occur at all (Fig. 2 in Williams & Scholl 2021a). Traps that have captured prey take on average about fourteen times as long to begin to reopen and seven times as long to fully reopen. Reopening of traps that have not captured prey is completed in about a day while those that have captured prey and digest it take weeks to reopen.

Table 1. Comparison of reopening times of traps that have captured prey (narrowed) with traps that have not.		
	Reopening Times (days)	
	No Capture	Capture (narrowed)
Number of Samples	10	5
Begin reopening (mean±SEM)	0.29±0.027	9.2±1.47
Fully reopened (mean±SEM)	0.90±0.038	12.8±1.92

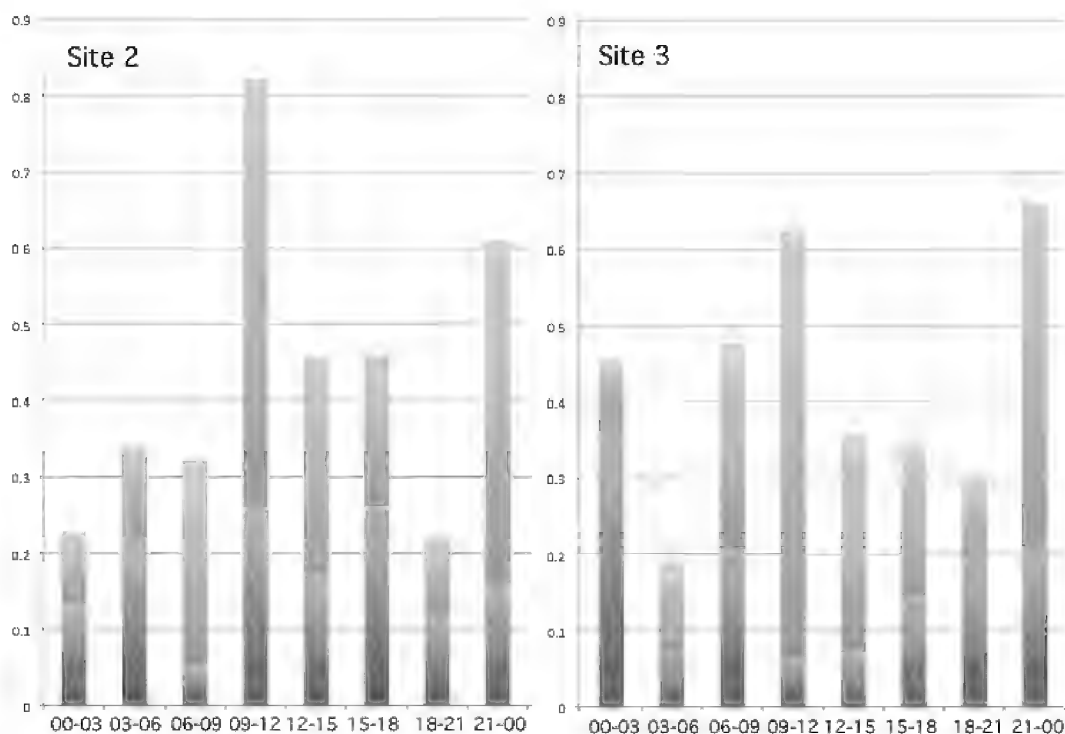


Figure 2: a. Ten-day average of captures and closures during 8 time periods at Site 2. Blue bars show the capture frequency. Red bars show the frequency of closure where there was no capture; the total bar height is the frequency of closures. The times at the base of the bars are 24 hours EST. The units on the vertical axis are closures/active trap/3 hours. b. Five-day average captures and closures during 8 time periods at Site 3. Blue bars show the capture frequency. Red bars show the frequency of closure where there was no capture and the total bar height is the frequency of closures. The times at the base of the bars are 24 hours EST. The units on the vertical axis are closures/active trap/3 hours.

Closure and capture frequencies

Frequencies of closure and prey capture of traps of two stands of *Dionaea* growing in the Gulf coastal plain in the Florida panhandle were determined from time-lapse photography. Site 2 is typical grassy coastal savannah amidst mature pine trees while Site 3 is a wetter area with *Sphagnum* near a stream within several meters of a stand of Titi trees (*Clitonia monophylla*).

The average frequency of prey capture, during 10 days, for traps at Site 2 was 1.39% of active traps/day and at Site 3 the average frequency, during 5 days, was 1.33% of active traps/day (Table 2).

Total trap closure, including closures that captured prey, had an average frequency of 3.45% of active traps/day at Site 2 and an average of 3.06% of active traps/day at Site 3. About 40% of the closures at both sites resulted in prey capture (Table 2). The highest capture frequency per day was 4.9% of active traps at Site 2. On that day 7.0% of the active traps closed and 70% of the closures resulted in captures.

The capture frequency of *Dionaea* in the field is low. The average frequency during the 15 days of observation was less than 1.4% of active traps capturing prey in a 24-hour period but when traps are triggered to close, they capture prey over 40% of the time. On the most successful day, 70% of closures resulted in prey capture. Traps are inefficient in luring prey to trigger the trap but are relatively effective in capturing the prey that does trigger trap closure. Some insects, spiders, snails, and even toads that are all far too large to be captured often trigger traps that close but fail to capture

Table 2. Trap closures per active-trap per day and closures with and without capture at two different sites.		
Location	Site 2	Site 3
Number of days sampled	10	5
Percent total closures (Clos/ActTr/Day)	3.45	3.06
Percent close no-capture (Clos/ActTr/Day)	2.07	1.73
Percent captures (Captures/ActTr/Day)	1.39	1.33
Percent closed that captured	40.3	43.5

prey. Both animals that are captured and those too large or strong for traps to capture blunder into traps. Their behavior is not what would be expected of an animal being lured by an attractant.

Comparison with previous research

The capture frequencies measured at both Florida sites were about half that reported in North Carolina in June by Williams (1980). Closure frequencies at both sites were lower than those reported in North Carolina. There were only one third as many closures at Site 2 and Site 3 as were measured in the Carolinas. The capture frequencies at Site 2 are one third to one half as many as in the earlier study in North Carolina, but of the traps that closed in the North Carolina study only 29.6% captured prey as opposed to greater than 40% in Florida.

Proportion of traps in open, narrowed, closed, and reopening states in summer and early fall.

When viewed in the field either in the Carolinas or in the Florida panhandle most traps appear to be open. Actual measurements of the proportion of traps that are open confirm this impression (Table 3). Eighty-three to ninety percent of the traps were open at each of five observation dates. Six to twelve percent of the traps had narrowed after capturing prey. Since traps that are narrowed often remain closed for a week or more it is not surprising that they are the largest group of traps that are not open. Less than 2% of the traps are closed and 2.4% to 5% of the traps are reopening either from the closed (with no capture) or the narrowed stage.

Table 3. Proportion of traps in various stages at noon EST during the summer and early fall of 2020. Total traps = active traps + narrowed traps. Reopening traps are reopening either from the narrowed stage or the closed stage. Traps were at Site 2 in a 15 × 24 cm area.					
Date	Traps Total	Percent Narrowed	Percent Closed	Percent Reopening	Percent Open
17-Jul	135	5.9	1.5	3.0	89.6
1-Aug	142	12.0	2.1	2.8	83.1
21-Aug	170	11.2	0.0	4.0	84.7
18-Sep	164	11.6	1.2	2.4	84.8
10-Oct	211	7.1	1.9	4.7	86.3

Closure and Capture frequency at different times of day

Closure and capture frequencies were measured for ten days at Site 2 and for five days at Site 3 during eight 3-hour time periods. *Dionaea* traps capture prey at all hours of the day. The capture frequencies were variable and were not significantly higher in any specific period. Closure of traps that did not capture prey also had variable frequencies with no relation to time of day (Fig. 2). Traps at Site 3 captured a few more prey at night. On this site we observed activity by nocturnal spiders that also were visible as remains in reopened traps. It is likely that any difference in frequency of closure or capture is due to the relative prevalence of nocturnal or diurnal prey instead of characteristics of the traps.

Interaction of traps with animals

Prey that were observed being captured included ants, gnats, spiders, and grasshoppers.

Ants, which typically compose about one third of the prey of *Dionaea* (Williams & Hartmeyer 2017), were observed being captured when scrambling from one place to another passing through several traps in the process. Occasionally they trigger a small or medium trap as they pass through and are captured. Most of the observed ants are small but a large (10 mm) ant was observed while in the center of one of the large traps away from the peripheral glands (Fig. 1).

Gnats often walk along the edge of traps pausing at the peripheral glands (aka “alluring glands”), that appear to act as nectaries (Fig. 3a). When behaving in this way they almost always move along the edge of the leaf, only rarely moving away from it. They usually walk from leaf to leaf but sometimes fly to an adjacent leaf. Gnats are only a small part of prey captured by *Dionaea* but they are sometimes captured when suddenly landing on a leaf (Fig. 3b). We observed small gnats and ants working the peripheral glands with their mouthparts in the same way as those reported by Jones (1923) and Hartmeyer *et al.* (2019). Gnats and ants feeding along the rim were not observed to cause trap closure even though both occasionally walked across the traps near the trigger hairs.

Peripheral glands are located along the rim of each trap lobe just outside the smooth area where the lobes seal during narrowing. Structurally they are nearly identical to the digestive glands



Figure 3: a. A 2.2 mm gnat gradually moves along the rim of the upper lobe of a trap pausing for extended periods, as it appears to feed on nectar from the peripheral glands. The sequence of video frames covers 30 seconds. b. A 3.5 mm gnat lands on a trap, brushes past the trigger hairs and is captured. Video frames from 3a and 3b are at the same magnification. 3b is black and white because the light intensity dropped below that required for color images.

that cover the inner surface of the lobes except for their smaller size and lower pigmentation. First described by Jones (1928) as “nectar glands?” that attracted small ants that did not set off the trap. They were named alluring glands by Lloyd (1942). Juniper *et al.* (1989) later called them the “peripheral glands”. The evidence for their role as nectaries were Jones’ observations and videos of captures made by the BBC (reviewed by Williams & Hartmeyer 2017; Hartmeyer *et al.* 2019). The name “alluring glands” suggest that these glands play a major role in attracting prey into the trap but there is little evidence that they do. We adopted Juniper’s term “peripheral glands” because it describes their position without designating a function.

Spiders typically compose up to a third of the prey of *Dionaea* (Williams & Hartmeyer 2017). Almost all of the spiders observed on traps were nocturnal wolf spiders. The smaller ones that are captured sometimes appear to be hiding in traps to avoid larger wolf spiders which have been observed stalking them. On one occasion a small spider was observed moving through the trap without being captured.

Grasshoppers are reported by Lichtner and Williams (1977) to compose about 15% of the prey they observed captured in a study conducted in June. These were probably smaller nymphs. Other studies, done at different times of year, report relatively few grasshoppers captured. A grasshopper, of medium size, was captured by a large trap (Fig. 4b) while eating its way along a grass blade that passed through the center of a trap. The leaf closed around it but the grasshopper was too large for the trap to form a seal between the two lobes. Later small ants were attracted to the leaf, some of which were captured by neighboring traps.

Potential prey that trigger traps then escape capture include beetles of both the weevil type (Fig. 3 in Williams & Scholl 2021) and the scarab type. Both are very strong and have a smooth hard

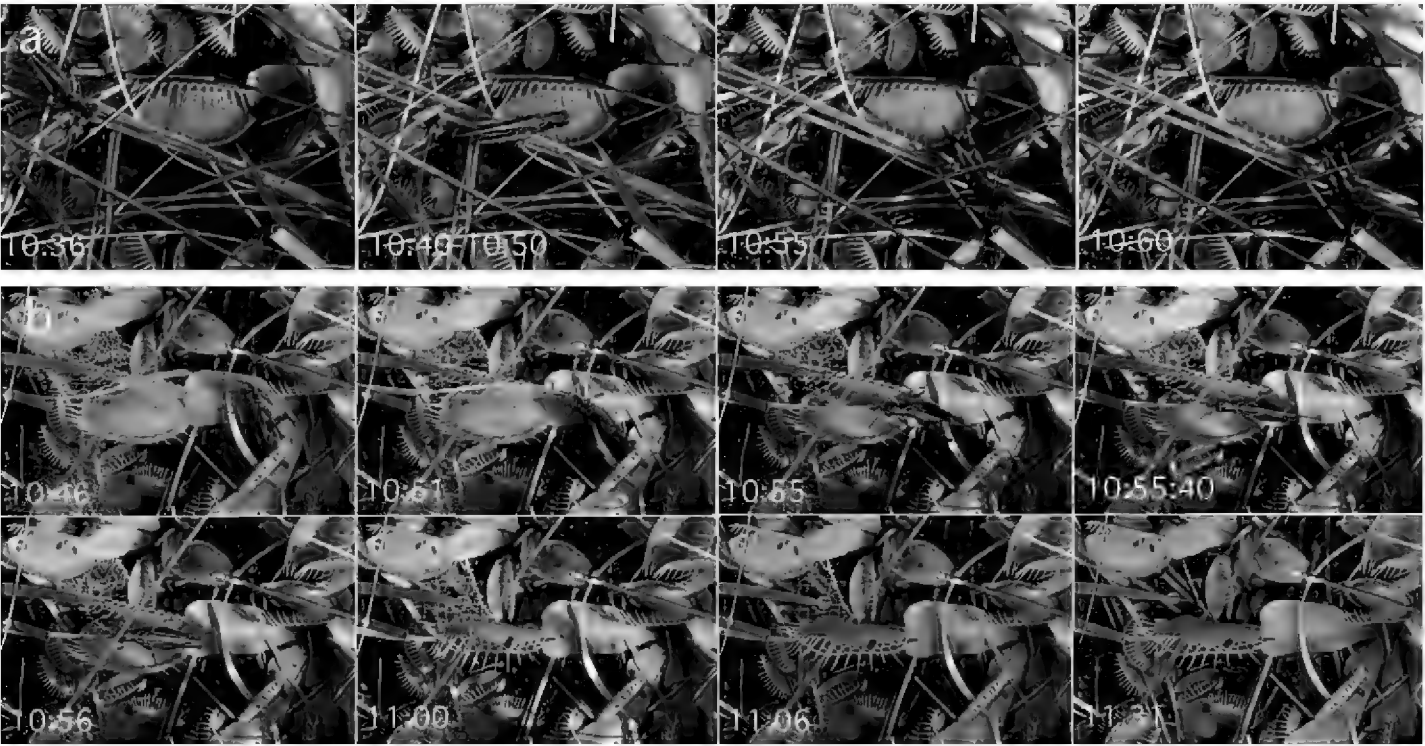


Figure 4: a. A grasshopper enters a *Dionaea* trap, remains there perfectly still for over 10 minutes and walks away without springing the trap. b. Simultaneously, a second grasshopper, feeding on a grass blade that passes through a trap, walks over the trigger hairs and triggers the trap resulting in its’ closing and narrowing. The relatively large grasshopper prevents the trap from sealing properly ultimately drawing ants to the partially closed trap.

carapace. In addition, the weevils have a pointed head and thorax that easily slips through the lobes of the trap. These insects can be cleanly captured, completely surrounded by the lobes of the trap for several minutes and slip out of its grasp and emerge in as little time as one minute, although one weevil struggled for 4 hours before breaking free. One weevil was trapped again and again setting off two additional traps within 5 minutes of its escape from the original trap.

Potential prey that enter traps and do not trigger them include ants, gnats, grasshoppers, and spiders. All have been observed entering traps and moving about without triggering closure. The ants and gnats avoid capture in the larger traps by keeping to the rim of medium and large traps where they are attracted away from the trigger hairs and toward the peripheral glands (Hartmeyer *et al.* 2019). In smaller traps they would easily reach the trigger hairs. It is possible that they serve as prey for smaller traps. On the same video as the grasshopper captured by a large trap (Fig. 4b) a second, nearly identical, grasshopper sat in the center of a large trap for over 10 min before exiting it without setting off the trap (Fig. 4a). Spiders sometimes crawl through traps without triggering them.

In preparing for this study, we saw a great many traps on plants that were cultivated in a terrarium catch ants and crickets in awkward ways. By contrast, in the captures we observed in the field, the trap lobes always surrounded the insect when triggered. Some beetles were able to escape after a clean capture.

Animals too large to capture that trigger traps were observed setting off traps. These include large grasshoppers, crickets and spiders, a narrow-mouthed toad, and a snail. These set off traps by stepping on them and sometimes by probing them. As many as three traps within 5 min were observed to be triggered by animals passing through the field of view. Small snakes, lizards, and an oak toad that are capable of triggering traps in this way have also passed through the field of view.

Large nocturnal wolf spiders often walk into the field of view and step on traps while they wait for prey, occasionally triggering them (Fig. 5). During the day, large grasshoppers grazing on grass also occasionally trigger traps. An unexpected visitor was a land snail that stuck its head into two

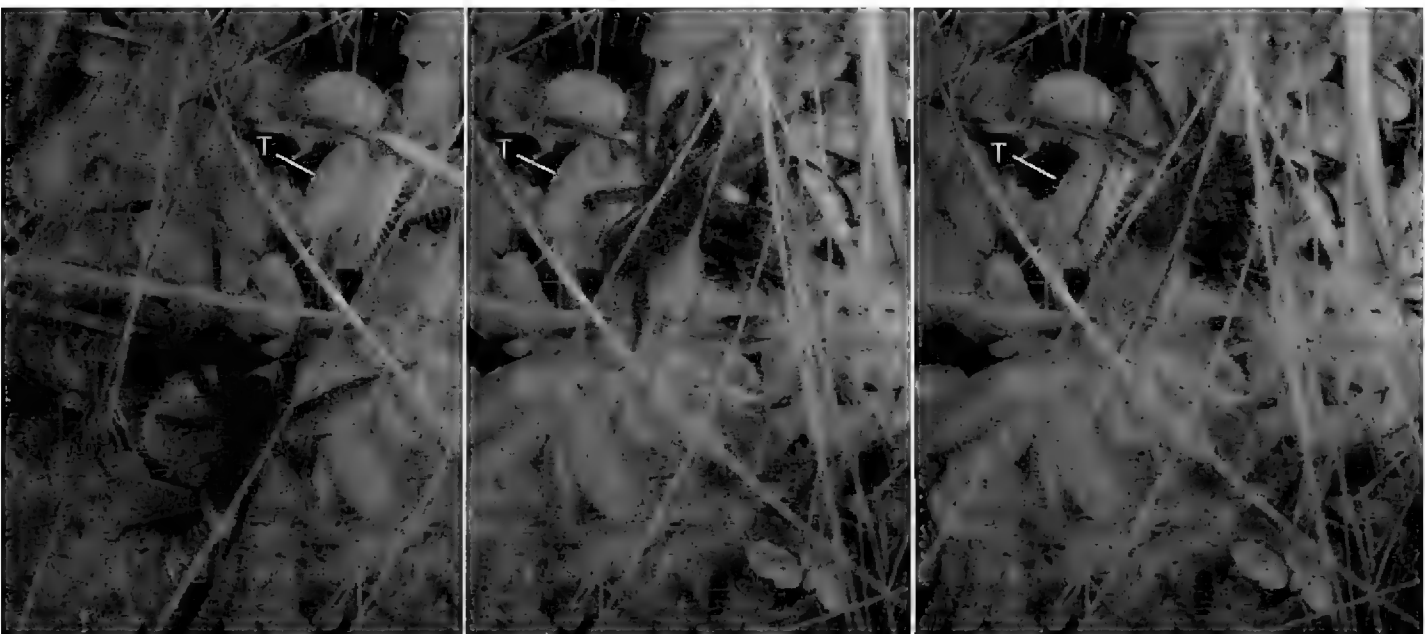


Figure 5: A large wolf spider steps on a trap “T” as it walks through a cluster of *Dionaea* plants just after midnight. Trap “T” is triggered but it has no chance of catching such a large spider.

traps, triggering their closure before pulling its head free from the closed trap (Fig. 1a). Many of the 60% of traps that close without capturing prey may be triggered in this way.

Efficient capture of the few potential prey that enter a trap

When *Dionaea* traps are triggered to close, about 40% of the closed traps capture prey yet fewer than 3.5% of the traps capable of capturing prey make captures on an average day. The capture mechanism is remarkably effective in catching visitors that trigger the trap but it does very poorly at attracting prey, suggesting that any postulated attractants the traps produce to attract prey are ineffective or absent. Yet, the peripheral glands do seem to attract small ants and gnats that move about the rim of the traps without triggering them. What is the function of the peripheral glands if they do not contribute to the capture of prey? In this study, we observed medium to large traps of mature plants. It is possible that in seedlings and the small traps of immature plants, the gnats and small ants provide important nutrients.

Summary

In a population of *Dionaea* in the field, either in the Carolinas or in Florida, most of the traps are open at any given time, allowing maximum exposure of the leaf surface to the sun. Capture and digestion of prey have been shown to supply mineral nutrients to the plants (Adamec 1977). The *Dionaea* plants appeared to be very healthy despite the fact that prey capture by individual traps is very low, indicating that a frequency of capture of 1.4% of the active traps per day in a population is adequate to provide the nutrients obtained from carnivory. The elaborate mechanism that closes the trap is remarkably effective at capturing animals that trigger the trap. As many as 40% to 44% of traps that are triggered to close capture prey.

Acknowledgements: We thank Dr. Susan Verhoek of Lebanon Valley College for help in framing the study and constructive comments on the research. We would like to thank Rick Williams of RC Williams Company for designing the camera holder that played a crucial role in this research.

References

- Adamec, L. 1997. Mineral nutrition of carnivorous plants: A review. *The Botanical Review* 63(6): 273-299.
- Brown, W.H., and Sharp, L.W. 1910. The closing response in *Dionaea*. *Bot. Gaz.* 49: 290-302.
- Lloyd, F.E. 1942. *Carnivorous Plants*. Chronica Botanica Co., Waltham, Mass.
- Lichtner, F.T., and Williams, S.E. 1977. Prey capture and factors controlling trap narrowing in *Dionaea* (Droseraceae). *American Journal of Botany* 64(7): 881-886.
- Hartmeyer, S.R.H., Hartmeyer, I., and Williams, S.E. 2019. *Dionaea* traps selectively allow small animals to escape. *Carniv. Pl. Newslett.* 48(4):153-160.
- Jones, F.M. 1923. The most wonderful plant in the world. *Natural History* 23(6): 589-596.
- Juniper, B.E., Robins, R.J., and Joel, D.M. 1989. *The Carnivorous Plants*. Academic Press, London.
- Pavlovic, A., and Mithöfer, A. 2019. Jasmonate signalling in carnivorous plants: copycat of plant defence mechanisms. *Journal of Experimental Botany* 70(13): 3379-3389.
- Scherzer, S., Böhm, J., Krol, E., Shabala, L., Kreuzer, I., Larisch, C., Bemm, F., Al-Rasheid, K.A., Shabala, S., Rennenberg, H., Neher, E., and Hedrich, R. 2015. Calcium sensor kinase activates potassium uptake systems in gland cells of Venus flytraps. *Proc. Natl. Acad. Sci.* 112(23):7309-7314.

Slack, A. 1980. Carnivorous Plants. MIT Press, Cambridge, Mass.

Williams, S.E. 1980. How Venus' flytraps catch spiders and ants. Carniv. Pl. Newslett. 9(3): 65, 75-78.

Williams, S.E., and Hartmeyer, S.R.H. 2017. Prey capture by *Dionaea muscipula*. A review of scientific literature with supplementary original research. Carniv. Pl. Newslett. 46(2): 44-61.

Williams, S.E., and Pickard, B.G. 1980. The role of action potentials in the control of capture movements of *Drosera* and *Dionaea*. In: Plant Growth Substances 1979 Edition: The Tenth International Conference on Plant Growth. Skoog, F. (ed.). pp. 470-480. Springer-Verlag, Berlin.

Williams, S.E., and Scholl, B. 2021a. Pollination of *Dionaea muscipula*, the Venus flytrap. Carniv. Pl. Newslett. 50(1): 16-23.

Williams, S.E., and Scholl, B. 2021b. Using a trail camera for recording plant-insect interactions with Venus flytrap as an example. Carniv. Pl. Newslett. 50(2): 66-72.

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NAPHTHOQUINONES IN PYGMY SUNDEWS (*DROSER*A SECT. *BRYASTRUM*)

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Keywords: *Drosera*, phytochemistry, naphthoquinones, chemotaxonomy.

Abstract: A detailed study of 44 accessions representing 38 taxa (76% of the diversity known at present) of pygmy sundews (*Drosera* sect. *Bryastrum*) reveals the first naphthoquinone patterns in this lineage, in which previous studies have not yielded reliable evidence for naphthoquinones. While most samples do not display detectable amounts of naphthoquinones as previously reported for the group, ramentaceone is detected in three mutually related taxa, and both ramentaceone and its regio-isomer plumbagin is present in all accessions investigated of *D. pulchella*.

Introduction

Pygmy sundews (*Drosera* sect. *Bryastrum*) are a well-defined lineage of ca. 50 species almost entirely confined to southwestern Western Australia (Fleischmann *et al.* 2018; Robinson *et al.* 2018). Only the more widespread *D. pygmaea* (reaching New Zealand) and the curiously disjunct *D. meristocaulis* (endemic to southernmost Venezuela) occur elsewhere. Almost all species (except the tropical *D. meristocaulis*) are known to form gemmae that are vegetative propagules from modified leaves (Goebel 1908; Rivadavia *et al.* 2013; Lowrie 2014: 44-58 & 116-151).

While naphthoquinones like plumbagin (2-methyljuglone, **P**) and ramentaceone (7-methyljuglone, **M**) have been known for a long time as characteristic, chemotaxonomically informative metabolites in the genus *Drosera*, they have not been detected with any certainty in pygmy sundews yet. A single, dubious report (reference to unpublished data without any experimental details) of **P** in *D. pygmaea* (Krenn in Länger *et al.* 1995) stands in clear contrast to negative results from a previous, comprehensive screening (Culham and Gornall 1994).

More recently, the re-investigation of species formerly believed devoid of naphthoquinones has yielded some unexpected, positive results (**P** in *D. menziesii*, Schlauer *et al.* 2018, **M** in *D. arcturi*, Schlauer *et al.* 2019a, and in *D. schizandra*, Schlauer *et al.* 2019b). The removal of ripe gemmae does not damage the rosettes that produced them, so this material was investigated for the first time in the present study.

Among numerous confirmed and new negative results, a few species are reliably found to contain **M**, and *D. pulchella*, previously considered devoid of naphthoquinones whatsoever, even contains both **M** and **P**.

Materials and methods

All plants used in the present study were raised from gemmae and cultivated at Andreas Fleischmann's greenhouse in southern Germany (Figs. 1 & 2). The geographic origin of all accessions was traced as far as possible (see Table 1). Ten to twenty fresh, ripe gemmae (ca. 10 mg) of each accession were harvested and investigated under the same conditions applying the same methods as

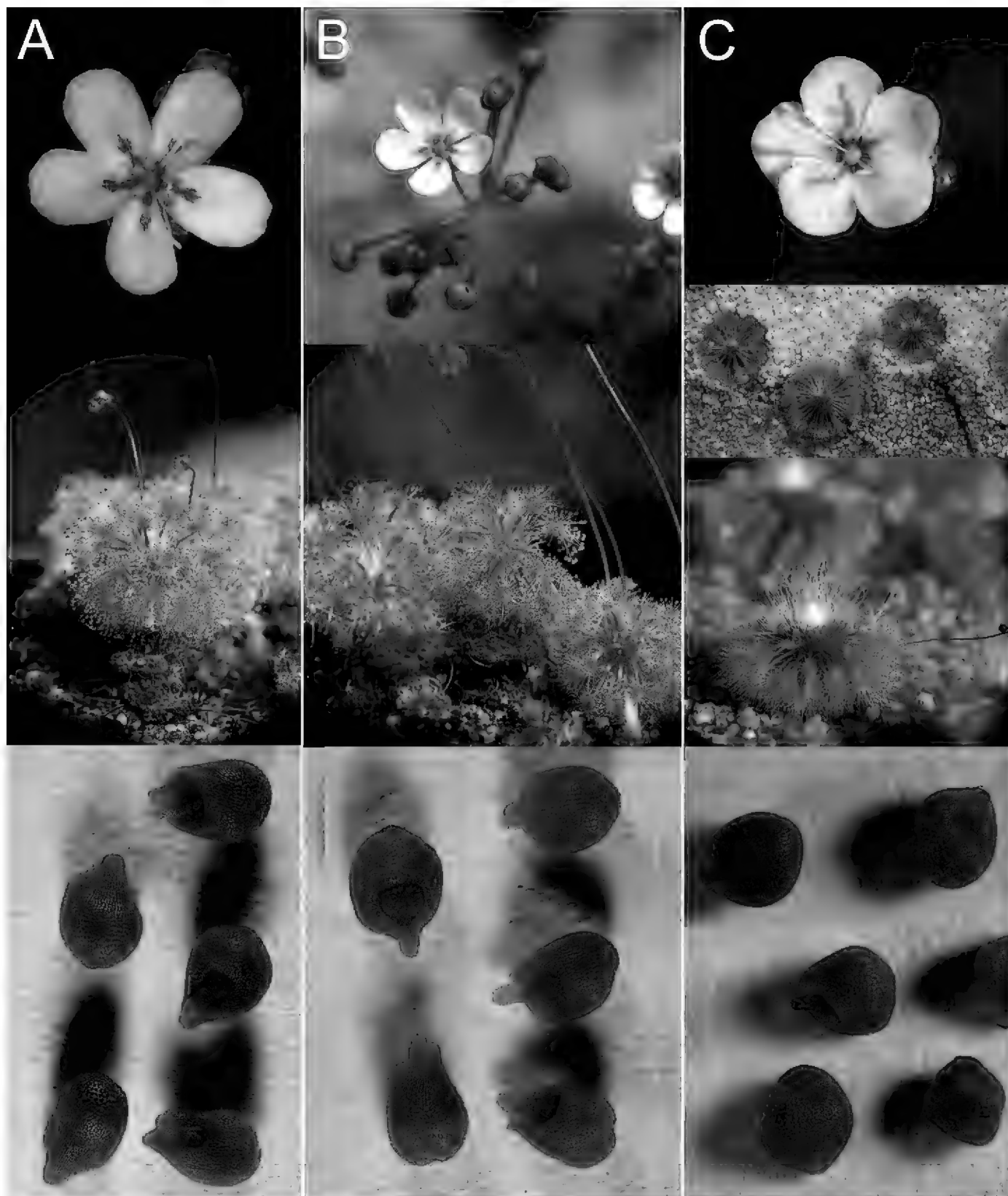


Figure 1: Flower, habit, and gemmae of the three M-positive members of *Drosera* sect. *Bryastrum*: A. *D. minutiflora*. B. *D. pedicellaris*. C. *D. sargentii*. Flower and habit images not at the same scale. Plants cultivated by A. Fleischmann, photo vouchers were made for the material used in this study.

reported previously for the investigation of leaves (Schlauer *et al.* 2017). Additionally, fresh leaves were investigated for some species or as control of the gemmae results.

Results

Naphthoquinones were detected in the investigated samples as summarized (together with previous results) in Table 1.

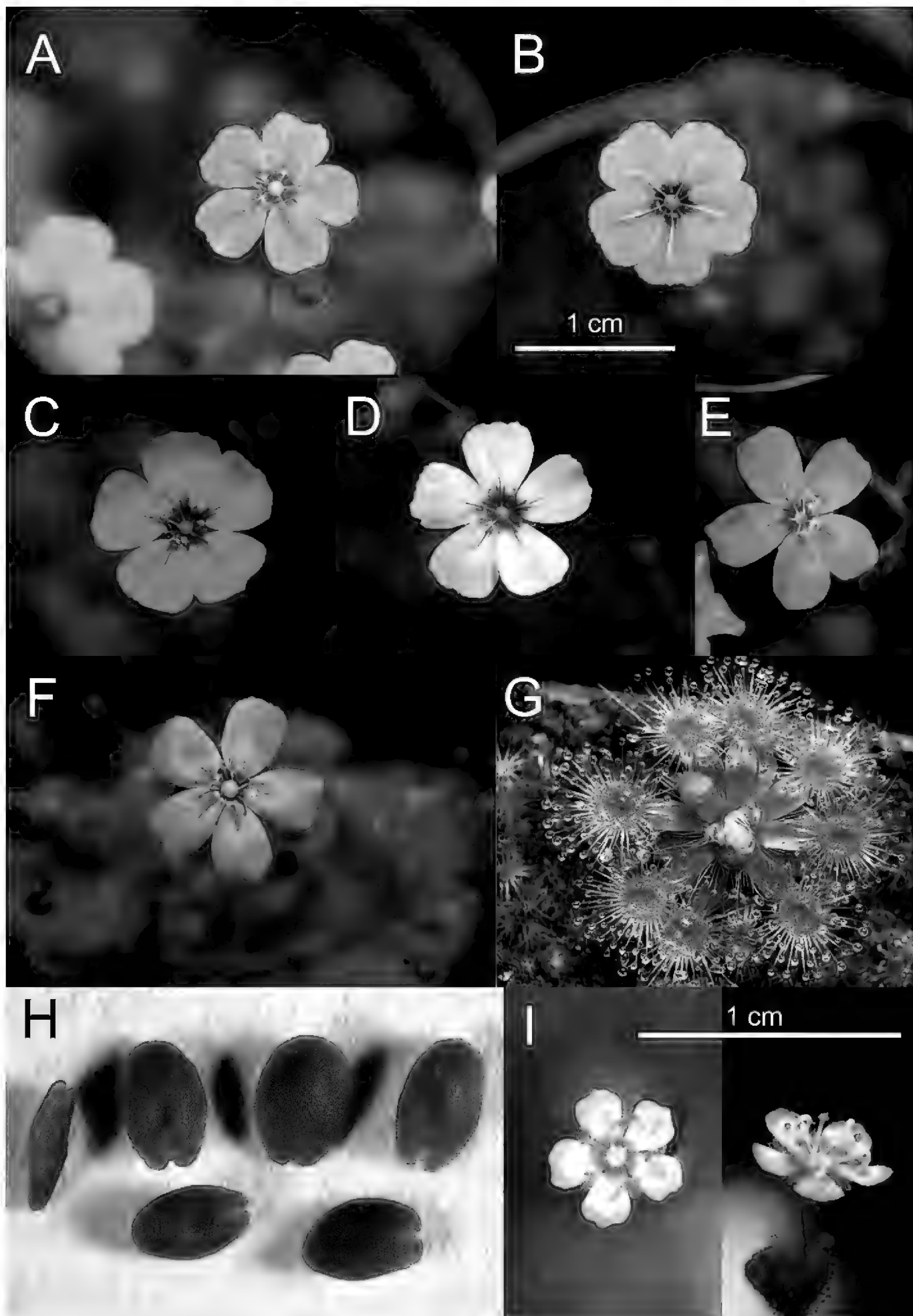


Figure 2: *Drosera pulchella* (A-H) and *D. depauperata* (I). Accessions of *D. pulchella* from Scott River, Western Australia (A, flower; H, gemmae), clone 52-A (B); Baker's Junction, Western Australia (C, flower; G, habit), white flower, red center (D), Mount Cooke, Western Australia (E), red flower (F). Images not at the same scale. Plants grown and photographed by A. Fleischmann.

Table 1. Taxa of <i>Drosera</i> sect. <i>Bryastrum</i> and quinones found.				
Taxon ^a	Accessions	Provenance	Quinone(s) ^b	References ^c
<i>D. androsacea</i>	1	Western Australia (WA)	0	*
<i>D. (occidentalis subsp.) australis</i>	1	WA	0	*
<i>D. barbigera</i>	1		0	1
<i>D. bindoon</i>	1	Bindoon, WA	0	*
<i>D. callistos</i>	1	Brookton, WA	0	*
<i>D. citrina</i>	1	Regans Ford, WA	0	*
<i>D. closterostigma</i>	1	(type form), WA	0	*
<i>D. coomallo</i>	1	Coomallo, WA	0	*
<i>D. depauperata</i>	1	locus classicus, WA	0	*
<i>D. echinoblastus</i>	1	WA	0	*
<i>D. enneabba</i>	2	Moore River NP, WA; (red from), WA	0	*
<i>D. gibsonii</i>	1	WA	0	*
<i>D. grieviei</i>	1	WA	0	*
<i>D. helodes</i>	2	Bullsbrook, WA; Great Northern Highway, WA	0	*
<i>D. hyperostigma</i>	1	WA	0	*
<i>D. lasiantha</i>	1	Porongorup NP, WA	0	*
<i>D. leucoblata</i>	1		0	1
<i>D. (nitidula var.) leucostigma</i>	1	WA	0	*
<i>D. mannii</i>	1	WA	0	*
<i>D. meristocaulis</i>	1	Sierra de la Neblina, Venezuela	0	2
<i>D. (occidentalis var.) microscapa</i>	1	locus classicus, WA	0	*
<i>D. miniata</i>	1	(type form), WA	0	*
<i>D. nitidula</i>	1		0	1
<i>D. (citrina var.) nivea</i>	1	WA	0	*
<i>D. occidentalis</i>	3		0	1
<i>D. (nitidula subsp.) omissa</i>	1		0	1
<i>D. (nitidula subsp.) omissa</i>	1	(pink flower), WA	M(+P-trace)	*
<i>D. oreopodion</i>	1	locus classicus, WA	0	*
<i>D. micrantha</i>	1		0	1 (as “ <i>D. paleacea</i> ”)

Table 1. Continued.				
Taxon ^a	Accessions	Provenance	Quinone(s) ^b	References ^c
<i>D. micrantha</i>	1	WA	0	*
<i>D. paleacea</i>	1	WA	0	*
<i>D. minutiflora</i> (?)	1		0	1 (as “ <i>D. parvula</i> ”)
<i>D. minutiflora</i>	1	WA	M	*
<i>D. (nitidula</i> var.) <i>patens</i>	1	WA	0	*
<i>D. (parvula</i> var.) <i>pedicellaris</i>	1	locus classicus, WA	M	*
<i>D. platystigma</i>	1		0	1
<i>D. pulchella</i>	2		0	1
<i>D. pulchella</i>	3	Scott River, WA; Baker’s Junction, WA; (white flower, red center), WA	P+M	*
<i>D. pulchella</i> × <i>nitidula</i>	1		0	1
<i>D. pulchella</i> × <i>occidentalis</i>	1		0	1
<i>D. pycnoblasta</i>	1		0	1
<i>D. pygmaea</i>	1		"P"	3
<i>D. pygmaea</i>	2		0	1
<i>D. pygmaea</i>	2	Windmill Lagoon, Tasmania; New Zealand	0	*
<i>D. (paleacea</i> subsp.) <i>roseana</i>	1	WA	0	*
<i>D. (parvula</i> subsp.) <i>sargentii</i>	1	WA	M	*
<i>D. scorpioides</i>	2		0	1
<i>D. scorpioides</i>	1	WA	0	*
<i>D. sewelliae</i>	1	(reddish form) WA	0	*
<i>D. (barbigera</i> subsp.) <i>silvicola</i>	1	WA	0	*
<i>D. (paleacea</i> subsp.) <i>stelliflora</i>	1	WA	0	*
<i>D. (paleacea</i> subsp.) <i>trichocaulis</i>	1	WA	0	*
<i>D. verrucata</i>	1		0	1 (as “ <i>D. dichrosepala</i> ”)
<i>D. verrucata</i>	1	Fishtrack Road, WA	0	*
<i>D. walyunga</i>	1	Walyunga NP, WA	0	*

^aNomenclature following Lowrie (2014), in parentheses alternative classification following Schlauer (1996).

^bP: plumbagin, M: ramentaceone, 0: no quinone detected

^cReferences

- * New/additional data from this study
- 1 Culham & Gornall 1994
- 2 Schlauer 2012
- 3 Länger *et al.* 1995

Fresh leaves of *D. roseana*, *D. scorpioides*, and *D. pygmaea* did not yield quinones, confirming gemmae and/or literature results. No quinones were likewise detected in leaf samples of *D. minutiflora* and *D. pedicellaris*, of which gemmae contained **M**. Quinones (**M** and **P**) in trace amounts were detected in leaves of *D. pulchella* (Mt. Cooke, red flower, Scott River).

Discussion

Although using gemmae instead of leaves should initially be only a test if less invasive sampling methods can be applied to delicate species, this study produced some noteworthy and chemotaxonomically relevant results. Previously, also seeds were found to contain naphthoquinones (unpublished results) but the amount required to yield reliable signals (usually the entire crop of an individual) cannot be expected to be available from most pygmy sundews, of which several taxa are more or less seedless for various reasons (e.g., self-sterility of the majority of species and usually only single clones available in cultivation, but also hybridity or aneuploidy, Kondo and Lavarack 1984).

The comparison between fresh leaves vs. gemmae confirm our assumption that gemmae are suitable for the investigation of quinone patterns. Especially where only tiny amounts of leaf material were available, gemmae provided more reliable data than leaves (“no detection” is just absence of evidence, not evidence of absence).

The discovery of **M** in *D. minutiflora* (syn. *D. parvula*: Lowrie 2014: 620), *D. pedicellaris*, and *D. sargentii* suggests a close phylogenetic relationship between these taxa as deduced from morphological similarity before (Schlauer 1996; Lowrie 2014: 700 & 796).

It is rather surprising that no quinone was found in *D. pulchella* and its hybrids before, as both **M** and **P** were clearly detected in the gemmae of all three accessions investigated in the present study. This widespread species is fairly isolated within *Drosera* sect. *Bryastrum* (Fleischmann, unpublished data) and none of the related taxa yielded any quinone so far. This likewise makes a hybrid origin of *D. pulchella* (that could be assumed from the chemotaxonomic pattern containing both quinones; see Schlauer and Fleischmann 2016) less probable.

The hypothesis that *D. depauperata* is conspecific with *D. pulchella* and just constitutes a depauperate form of it (Lowrie *et al.* 2017; Fleischmann *et al.* 2018) is not supported by the very different quinone content of both species, but it remains possible that the former is derived from a hybrid.

Drosera (*nitidula* subsp.) *omissa* is the only taxon in the *D. nitidula* group that contains quinones (predominantly **M** with traces of **P**) whatsoever. Only one accession was, however, investigated in the present study, and the group is remarkable for its well documented ability to hybridize even with phylogenetically more remote taxa (Lowrie 2014: 52-56), including the quinone-rich *D. pulchella*. Interestingly, Culham and Gornall (1994) did not detect quinones in any of the *D. pulchella*-hybrids investigated in their study. However, as pygmy *Drosera* were/are frequently misidentified in cultivation, also in botanical gardens and some carnivorous plant nurseries, any previously published data for this group from cultivated material (and without any voucher specimens) has to be taken with some care.

Acknowledgements: Johannes Betz, Thomas Carow, Dieter Kadereit, and Manfred Meisterl are thanked for sharing growing hints and gemmae of pygmy *Drosera*, some of them since more than two decades. Simon Poppinga and Siegfried Hartmeyer are thanked for reviewing and improving the manuscript.

References

- Culham A., and Gornall, R.J. 1994. The taxonomic significance of naphthoquinones in the Droseraceae. *Biochem. System. Ecol.* 22: 507-515.
- Fleischmann, A., Cross, A.T., Gibson, R., Gonella, P.M., and Dixon, K.W. 2018. Systematics and evolution of Droseraceae. In: Ellison, A.M. and Adamec, L. (eds.): *Carnivorous Plants: Physiology, Ecology, and Evolution*. Oxford University Press, 45-57.
- Goebel, K. 1908. Morphologische und biologische Bemerkungen. 18. Brutknospenbildung bei *Drosera pygmaea* und einigen Monokotylen. *Flora* 98: 324-335.
- Kondo, K., and Lavarack, P.S. 1984. A cytotaxonomic study of some Australian species of *Drosera* L. (Droseraceae). *Bot. J. Linn. Soc.* 88: 317-333.
- Länger, R., Pein, I., and Kopp, B. 1995. Glandular hairs in the genus *Drosera* (Droseraceae). *Plant. Syst. Evol.* 194: 163-172.
- Lowrie, A. 2014. *Carnivorous Plants of Australia: Magnum Opus*. Redfern Natural History Productions, Poole.
- Rivadavia, F., Miranda, V.F.O., Hoogenstrijd, G., Pinheiro, F., Heubl, G., and Fleischmann, A. 2012. Is *Drosera meristocaulis* a pygmy sundew? Evidence of a long-distance dispersal between Western Australia and northern South America. *Ann. Bot.* 110: 11-21.
- Robinson, A.S., Cross, A.T., Meisterl, M.E., and Fleischmann, A. 2018. A new pygmy sundew, *Drosera albonotata* (Droseraceae), from the western Wheatbelt and an updated diagnostic key to the orange-flowered pygmy *Drosera* of Western Australia. *Phytotaxa* 346: 221-236.
- Schlauer, J. 1996. A dichotomous key to the genus *Drosera* (Droseraceae). *Carniv. Pl. Newslett.* 25: 72-75.
- Schlauer, J. 2012. Literature reviews. *Carniv. Pl. Newslett.* 41: 121.
- Schlauer, J., and Fleischmann, A. 2016. Chemical evidence for hybridity in *Drosera* (Droseraceae). *Biochem. Syst. Ecol.* 66: 33-36.
- Schlauer, J., Hartmeyer, S.R.H., and Hartmeyer, I. 2017. Unexpected discovery of 7-methyljuglone (ramentaceone) in several Australian sundews. *Carniv. Pl. Newslett.* 46: 20-22.
- Schlauer, J., Fleischmann, A., and Carow, T. 2019a. Quinones from “Gondwanan” sundews. *Carniv. Pl. Newslett.* 48: 13-17.
- Schlauer, J., Hartmeyer, S.R.H., and Hartmeyer, I. 2019b. Chemistry and surface micromorphology of the Queensland sundews. *Carniv. Pl. Newslett.* 48: 111-116.

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AN ACCOUNT OF *DROSERA* SECTION *LASIOCEPHALA*

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Abstract: Over the past two decades interest has grown in the tropical *Drosera* of northern Australia. As more botanical research is conducted in this remote region, new species have been described, new undescribed taxa discovered and we have gained a greater understanding of their ecology. Lowrie (1998) provided the first detailed account of *Drosera* section *Lasiocephala*, additional accounts were published in Lowrie (2014) and Lowrie *et al.* (2017), since then further field work has been undertaken, phylogenetic studies (Fleischmann *et al.* 2018), and a new species has been described (Baleeiro *et al.* 2020). The aim of this paper is to synthesize the current body of knowledge into the most up to date summary of this section, and draws extensively on previous text created by the authors (Lowrie 1998; Bourke & Nunn 2012; Lowrie 2014; Lowrie *et al.* 2017), with the addition of new studies (Fleischmann *et al.* 2018; Baleeiro *et al.* 2020) and many new images.

Introduction

Drosera section *Lasiocephala* Planchon inhabits far northern Australia in a band that stretches from the Kimberley in the west through the northern part of the Northern Territory and northern Queensland, from north of Cairns to the tip of Cape York, extending across the islands of the Torres Strait and into southern coastal Papua New Guinea. This section comprises the perennial petiolaris complex species and two erect annual species, the type species for this section is *Drosera petiolaris* R.Br. ex DC.

Botanical history

Drosera is the largest carnivorous plant genus with ca. 250 species known worldwide, 147 of which are endemic to Australia (Lowrie 2014; Fleischmann *et al.* 2018). The majority of *Drosera* species are perennial herbs, including 14 of the 16 currently recognised species of *Drosera* section *Lasiocephala*, the other two being annual, which sits within *Drosera* subgenus *Ergaleium* per the infrageneric classification of Fleischmann *et al.* (2018).

The first species described from *Drosera* section *Lasiocephala* were *Drosera petiolaris* R.Br. ex DC. and *D. banksii* R.Br. ex DC in 1824 (de Candolle 1824). The type specimens of *D. petiolaris* and *D. banksii* were collected in 1770 by Banks and Solander on the Captain James Cook's voyage aboard the HMS Endeavour. They were collected at the Endeavour River, Cooktown, Queensland. *Drosera fulva* Planch. was described in 1848, from a collection by John W. Armstrong sometime between 1838 and 1840, in the region of Port Essington (175 km northeast of Darwin). Diels (1906) treated *D. fulva* as a synonym of *D. petiolaris* and subsequent authors supported this until Lowrie reinstated this species in 1998 (Lowrie 1998). Marchant and George (1982) described *D. subtilis* in Flora of Australia, from material collected in 1980 from the Mitchell Plateau. Katsuhiko Kondo (1984) formally described three closely related additional taxa, *D. dilatatopetiolaris*, *D. falconeri*, and *D. lanata*. Lowrie undertook a significant amount of research into the *D. petiolaris* complex in the 1990's and described a further eight species from across northern Australia (Lowrie 1994,

1996a, 1996b, 1997, 1998, 2014). The most recent addition to *Drosera* section *Lasiocephala* was in 2020 when *D. stipularis* was described from Far North Queensland (Baleeiro *et al.* 2020).

The section name *Lasiocephala* is derived from the Greek *lasios* (shaggy) and *kephale* (head), a reference to densely hairy growing points. These species were originally placed in *Drosera* subgenus *Rorella* sect. *Lasiocephala* Planch. (Planchon 1848). In a recent classification, section *Lasiocephala* was included under subgenus *Drosera* (Seine & Barthlott 1994), however Fleischmann *et al.* (2018) placed this section in *D.* subgenus *Ergaleium*, which includes the majority of Australian taxa, and this placement is supported in this paper. The erect annual species *D. banksii* and *D. subtilis*, were formerly included in *D.* section *Annuerecta* Lowrie (Lowrie 2014) and sect *Ergaleium* (Marchant & George 1982) are here included in *D.* section *Lasiocephala* on account of new phylogenetic data (Fleischmann *et al.* 2018) supporting this placement.

Species roster – etymology, distribution, habitat, and phenology

There are currently 16 published species in *Drosera* section *Lasiocephala* and at least 8 new species awaiting formal description (Lowrie 2014; Lowrie *et al.* 2017; Baleeiro *et al.* 2020).

Drosera banksii R.Br. ex DC (Fig. 1A)

The epithet *banksii* honours Sir Joseph Banks (1743-1820), a famous patron of the natural sciences and President of the Royal Society for over 40 years who sailed around the world with Captain James Cook (1768-1771). *Drosera banksii* has a northern distribution in Western Australia (Packhorse Range; Beverley Springs Station); Northern Territory (Humpty Doo; Kakadu) and Queensland (Endeavour River). It is also recorded from western Papua New Guinea. This species grows in red loam by creeks, on sandy creek banks in black sand, and on watersheds near sandstone rock outcrops. This species flowers from May to July.

Drosera brevicornis Lowrie (Fig. 1C)

The epithet *brevicornis* is derived from the Latin *brevis* (short) and *cornis* (horned), in reference to the horn-like apical filament projection above the anthers. Mainly known from Northern Territory where, on the mainland, *D. brevicornis* is recorded from the Darwin region, notably Palmerston to Batchelor, south of Darwin, and eastwards towards and including Kakadu National Park. In Queensland, it is known from Sweers Island in the Gulf of Carpentaria. *Drosera brevicornis* grows on gravel slopes in hilly areas, or in shallow depressions in flat country, predominantly in the slower moving watershed zones where sand and loam particles can accumulate amongst the gravel. This species flowers from March to April.

Drosera broomensis Lowrie (Fig. 1D)

The epithet *broomensis* refers to the Broome region in the Kimberley, Western Australia, where this species occurs. *Drosera broomensis* is known from the Kimberley, Western Australia, where it is apparently confined to the Dampier Peninsula. This species is commonly found in soils that also have an association with beige coloured termite mounds. It grows in sandy soils amongst thinly spread, annual cane grass (*Mnesithea rottboellioides*) on bare patches of ground that border seasonally wet depressions and lake shores to the north and northeast of Broome. This species flowers from February to March.

Drosera caduca Lowrie (Fig. 1E)

Drosera caduca is named from the Latin *caducus* (fallen or liable to fall), a reference to the traps that are present only on the early season growths, but reduced and eventually absent on all subse-

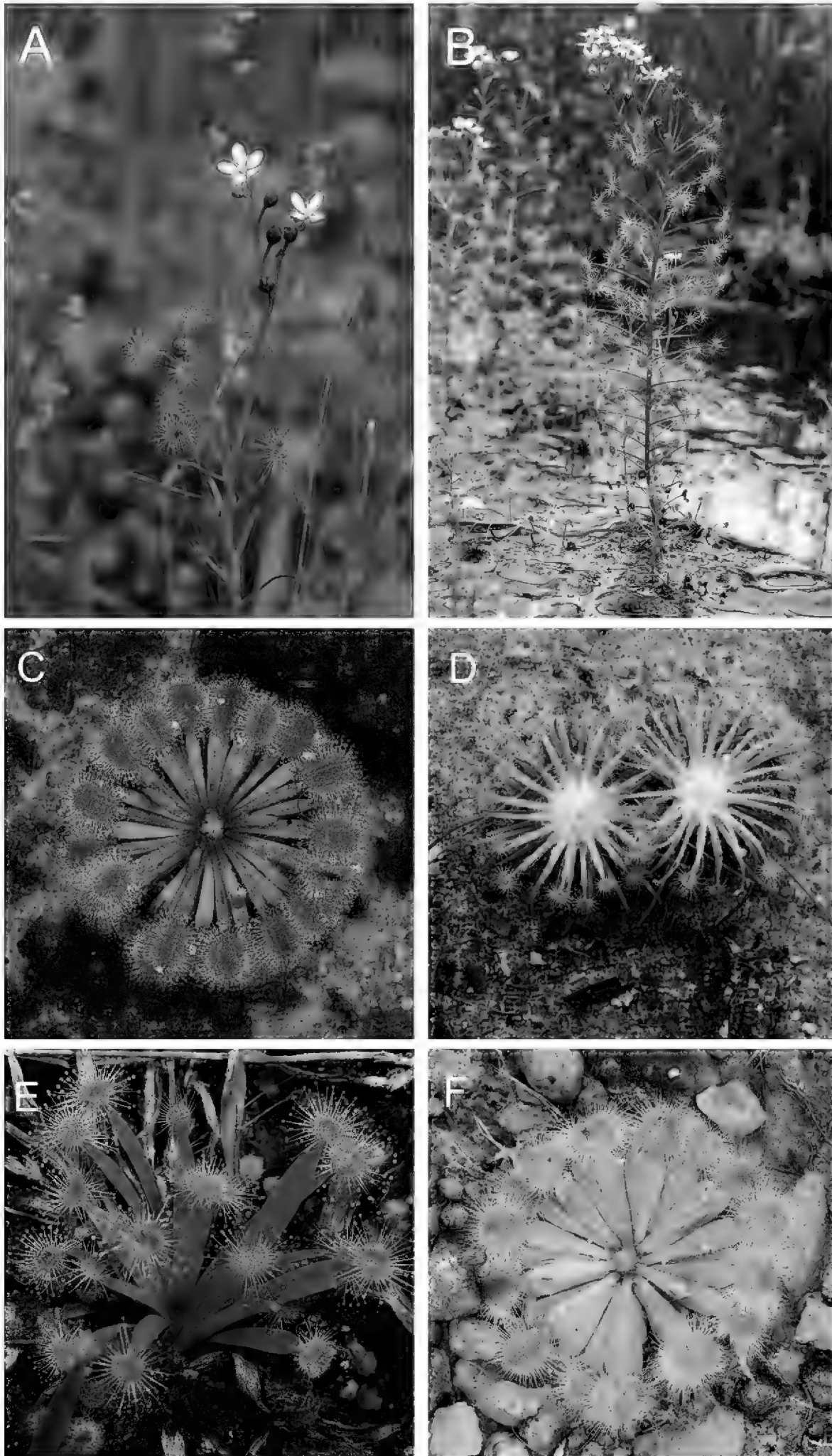


Figure 1: A: *Drosera banksii*; B: *D. subtilis*; C: *D. brevicornis*; D: *D. broomensis*; E: *D. caduca*; F: *D. darwinensis*. Photos: A, C, F - R. Nunn; B - A. Robinson; C - G. Bourke; E - A. Lowrie.

quent leaves. *Drosera caduca* is distributed throughout the Edkins Range to the southern regions of the Prince Regent River Reserve. It also occurs on Augustus Island, Kimberley, Western Australia. On the mainland, it grows on creek margins in silty white sand soils. On Augustus Island, it grows in stony brown sand on the edge of a basin on the lower slopes of a sandstone ridge with *Eucalyptus miniata* woodland and *Spinifex*. This species flowers from December to July.

Drosera darwinensis Lowrie (Fig. 1F)

The epithet *darwinensis* refers to the Darwin region of the Northern Territory, where this species occurs. *Drosera darwinensis* is distributed from Palmerston to Berry Springs, south of Darwin, and eastwards to Humpty Doo in the Northern Territory. It grows in clayey-sand with laterite pebbles. This species flowers in December to April

Drosera derbyensis Lowrie (Fig. 2A)

The epithet *derbyensis* refers to the Derby region in the Kimberley, Western Australia, where this species was discovered growing near the Boab Prison Tree, near Derby, in 1988. *Drosera derbyensis* is known from Western Australia and is a common species on the Yampi Peninsula, as well as on a number of the off-shore islands in the Buccaneer Archipelago to the northwest of the peninsula. *Drosera derbyensis* also extends south of the Yampi Peninsula to the Erskine Ranges, and to the southeast to the Leopold Ranges. *Drosera derbyensis* grows in sandy soils on the aprons of sandstone rock outcrops, as well as in seasonally wet savannahs. This species flowers in March to June.

Drosera dilatatopetiolaris K.Kondo (Fig. 2B)

The epithet *dilatatopetiolaris* is from the Latin *dilatatus* (enlarged, widened) and *petiolus* (petiole), in reference to the wider leaves of this species in comparison to the narrower leaves of *Drosera petiolaris*. This species ranges widely from the Kimberley in Western Australia and east to the Darwin region. There are official records from Beverley Springs and King Edward River, while in Northern Territory there are records from Palmerston and Humpty Doo. This species grows in sandy soils that are damp but not flooded during the wet season, favouring sandy flats near sandstone outcrops, *Eucalyptus* savannahs, and open mixed woodlands. This species flowers in October.

Drosera falconeri K.Kondo (Fig. 2C; Front Cover)

The epithet *falconeri* honours Mr. D. Falconer, a tropical aquarium fish collector in the Northern Territory, who first discovered this species whilst on a fish-collecting expedition to the Finnis River. This species occurs in Northern Territory, with records from Palmerston, Berry Springs, Fly Creek, the Finnis River catchment, and Melville Island. It grows in grey, silty clay, often on gently sloping savannah in areas that are inundated for periods of time, especially during the earlier part of the wet season. This species flowers in November to December.

Drosera fulva Planch. (Fig. 2D; Back Cover)

The epithet *fulva* is from the Latin *fulvus* (reddish yellow or tawny brown), in reference to the colour of the pressed herbarium specimens on the type sheet. Records of this species all come from the Northern Territory, including Howard Springs, Palmerston, Humpty Doo, and Port Essington on the Cobourg Peninsula. It grows in sandy soil on damp flats, seepage areas and ephemeral wet depressions just above the wet season flood levels. If heavy rains and flooding are experienced, plants may remain under water for several weeks at a time. This species flowers in February to May.

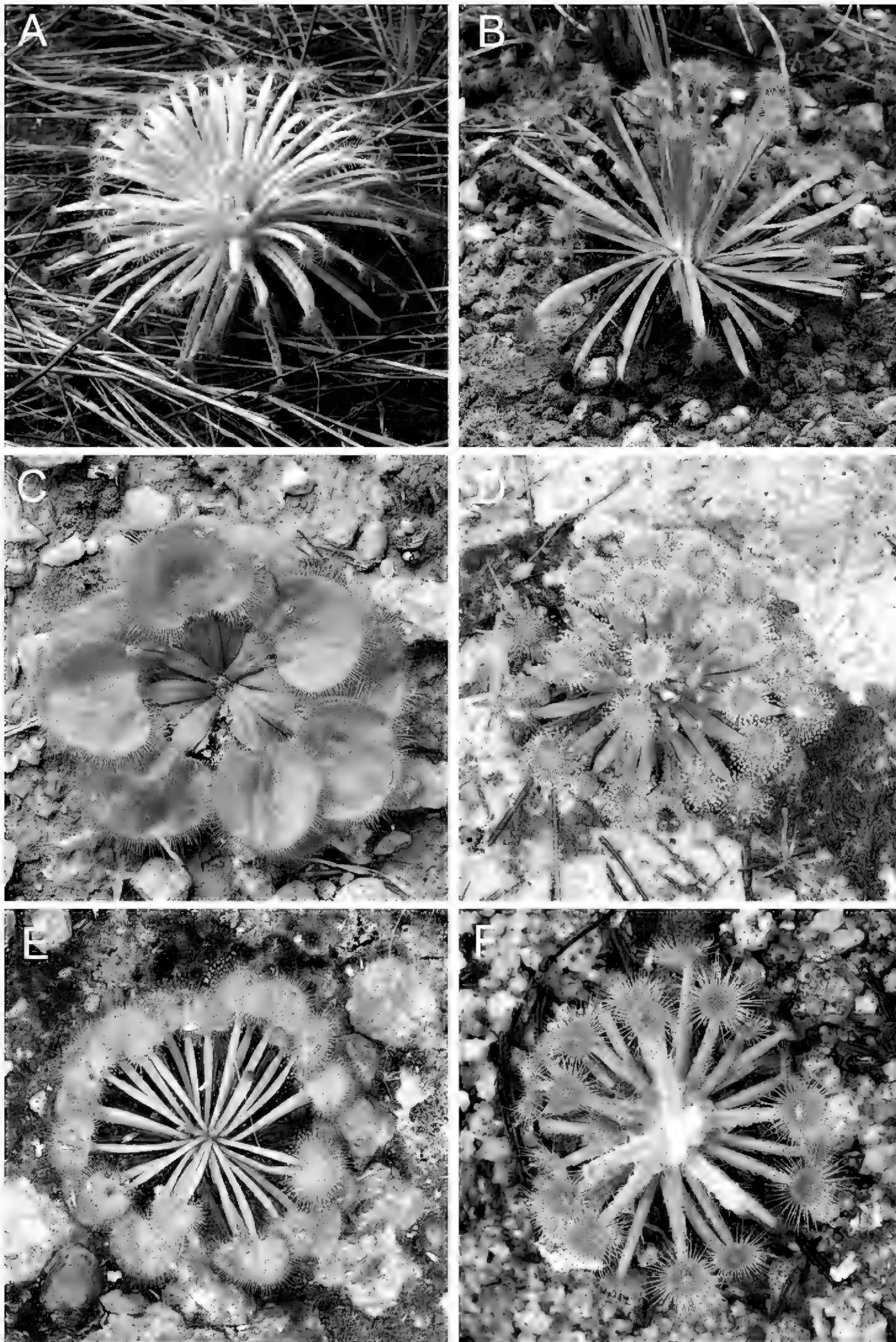


Figure 2: A: *Drosera derbyensis*; B: *D. dilatatopetiolaris*; C: *D. falconeri*; D: *D. fulva*; E: *D. kenneallyi*; F: *D. lanata*. Photos: A – G. Bourke; B-F – R. Nunn

Drosera kenneallyi Lowrie (Fig. 2E)

The epithet *kenneallyi* honours Professor Kevin Francis Kenneally, Botanist and Principal Research Scientist, who first collected this species on 7 December 1982. *Drosera kenneallyi* is known from Western Australia, with records from the Mitchell Plateau area of the Kimberley region, and the Northern Territory, with records from Dundee Beach. The type material was collected in skeletal soil over hardpan laterite near Airfield swamp. In the Northern Territory it grows in silty sand soils over laterite in association with grey, magnetic termite mounds. This species flowers in November to early December.

Drosera lanata K.Kondo (Fig. 2F)

The epithet *lanata* is from the Latin *lanatus* (woolly), a reference to its very dense, woolly indumentum. *Drosera lanata* is endemic to Queensland, with records from Mareeba, Chewko, and Undarra. It grows in sandy or gravelly well-drained soils in woodlands or in shallow, decomposed granite in rocky areas, often in open *Casuarina* woodland. In the Mareeba region on the Atherton Tablelands, it favours well-drained white sand habitats. This species flowers in January to March.

Drosera ordensis Lowrie (Fig. 3A)

The epithet *ordensis* refers to the alluvial valley of the Ord River, in which this species grows. *Drosera ordensis* is known from Western Australia, with records from Kununurra, Wyndham, Mount Elizabeth Station, and Pago, as well as the Northern Territory (Keep River National Park). *Drosera ordensis* grows in sandy soils derived from sandstone rock. Large colonies of plants are commonly found near banded sandstone outcrops. This species flowers in December to April.

Drosera paradoxa Lowrie (Fig. 3B)

The epithet *paradoxa* is from the Greek *paradoxos* (contrary to expectation), referring to the complicated and poorly understood growth cycle of this species, which was only revealed after considerable field study over a number of years during the wet and dry seasons. *Drosera paradoxa* is widely distributed in Western Australia from the west and north coasts of the Kimberley, inland to Beverley Springs, and eastwards to Arnhem Land and Kakadu National Park in the Northern Territory. *Drosera paradoxa* grows in skeletal, sandy soils over sandstone pavements on the banks, as well as beds, of seasonally dry creeks, in the cracks of sandstone pavements, in pads of moss adjacent to sandstone seepages, as well as amongst sandstone boulders. Flowers March to May.

Drosera petiolaris R.Br. ex DC (Fig. 3C)

The epithet *petiolaris* is from the Latin *petiolus* (petiole) and *-aris* (provided with), in reference to the long, narrowly petiolate leaves of this species. This species is recorded from Queensland (Endeavour River to Cooktown, Musgrave, Sanamere, Lockhart River) and the southern coastal plain of the island of New Guinea. It grows in sand near creeks and pools, particularly around sedge, as well as open wet scrub. It is commonly found standing in shallow water during the wet season. This species flowers in October to April.

Drosera stipularis Baleeiro, R.W.Jobson and R.L.Barrett (Fig. 3D)

The specific epithet refers to the erect to spreading stipules exerted above the central rosette. Known only from two collections north of Cooktown and east of Starke National Park, on Cape York Peninsula, Queensland. Found on moist pockets (each pocket c. 20 m²) of fine sand substrate within low heath and sedges. Found in flower in April and July.

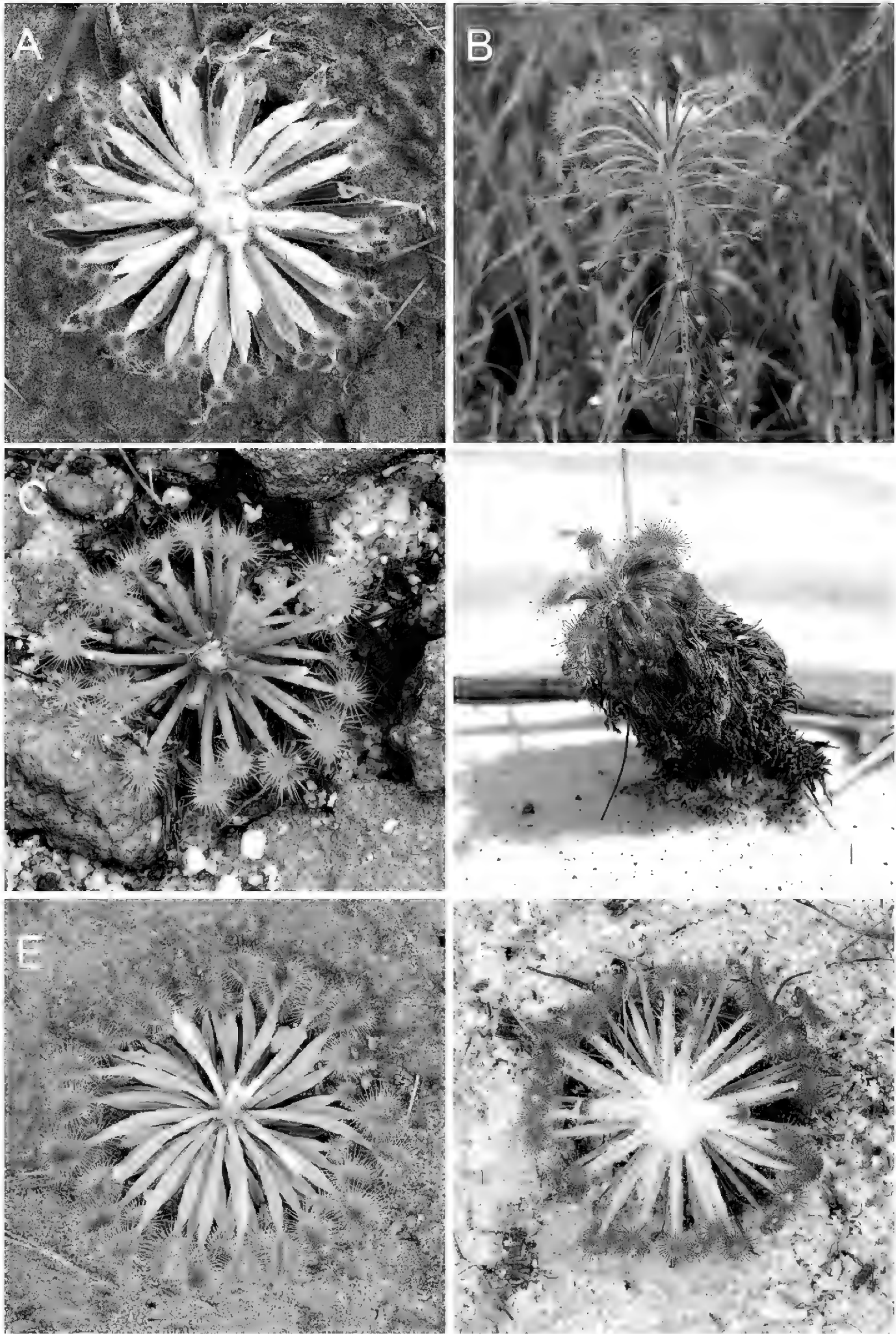


Figure 3: A: *Drosera ordensis*; B: *D. paradoxa*; C: *D. petiolaris*; D: *D. stipularis*; E: *D. aff. brevicornis* from Kimberley; F: *D. aff. lanata* from NT. Photos: A,B,C,E,F – R. Nunn; D – P. Baleeiro.

Drosera subtilis N.G.Marchant (Fig. 1B)

The epithet *subtilis* is derived from the Latin *subtilis* (fine, delicate), in reference to its somewhat slender overall form. *Drosera subtilis* is known from Western Australia, where it is widespread throughout the Kimberley region, with records from Beverley Springs Station, Bigge Island, the Mitchell Plateau (Camp Creek), and Theda Station. It has also been recorded in the Northern Territory (Little Nourlangie Rock). It grows in skeletal soils of sand and black humus over sheet sandstone, in areas covered with a film of water such as that found on the margins of seepage lines. It also grows in wet, black mud on the edges of sheet basalt, and in gravel in the seepage lines of basalt pavements. This species flowers in October to April.

Undescribed Taxa

Lowrie (2014) included another eight new taxa that are currently in the process of being described. Many of these are now in cultivation and readers may ask why they haven't been formally described by now. Describing plants from cultivated specimens is a poor practice, as it will never lead to a description that accurately expresses the variation of the species exhibited in the field. The use of cultivated specimens for type specimens should also be discouraged for the same reason. The naming of new species should rest with those botanists who collect specimens and study plants in the field as well as in the herbarium. It is only in the plant's natural habitat that the biology of the species can be closely examined and educated judgments be made about its uniqueness. Once the field data have been examined and comparisons made with known species, the process of naming a new species can properly begin. The naming of new species is time consuming and arduous. The task must be done thoroughly in order to have any scientific merit. Specimens of related species must be examined and much of this material is difficult to obtain from the field. The complete life cycle of each proposed species must be examined so that comparisons can be made with its nearest relatives. The majority of the northern regions of Australia are remote and lack roadways, and travel in the wet season in most areas is logistically impossible without the aid of a helicopter. The northern regions of tropical Australia are poorly explored botanically, and it is certain that additional new species will continue to be discovered in the coming years. These eight taxa are as follows.

Drosera species from Bigge Island (Fig. 4D)

This tiny, prop-rooted, rosetted species bearing long peduncles and a many-flowered rachis is yet another example of a new species being collected from a region that has previously been poorly botanically explored. It was discovered during a botanical survey on the uninhabited Bigge Island off the Australian mainland and growing in sandy soils between *Triodia* (Spinifex) species.

Drosera species from Tanami Desert (Fig. 4F)

This is an arid growing, hairy leaved, white flowered, 50 mm diameter, basal rosetted species from the Tanami Desert in the Northern Territory and the adjoining Great Sandy Desert region of Western Australia. It grows in the open sandy soils between *Triodia* species (Spinifex bushes) and the *Acacia* species shrubs of these desert regions. Although they are deserts in name, they do still receive their fair share of rain events at different times of the year via thunderstorms as well as regular morning fogs.

Drosera aff. *brevicornis* from Kimberley (Fig. 3E)

A large, compact, wide leaved, rosetted species with large white flowers that develop huge mature and immature seed capsules. Known from the Mitchell Plateau, Theda, as well as occurring off shore on Augustus Island in the Kimberley region of Western Australia.

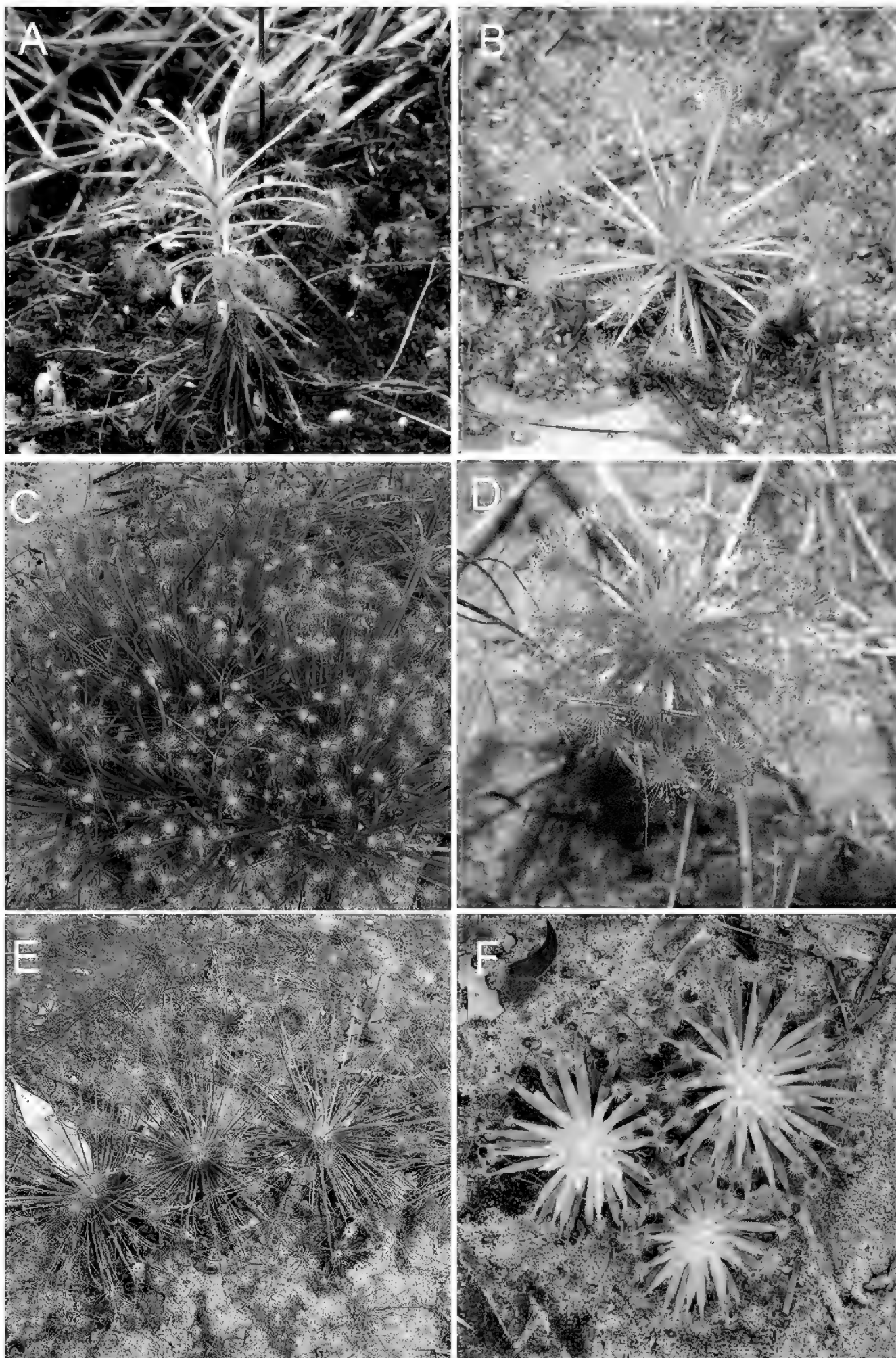


Figure 4: A: *Drosera* aff. *paradoxa* (Orange Flower) NT; B: *D. aff. paradoxa* (Swamp Form); C: *D. aff. petiolaris* from NT; D: *D. species* from Bigge Island; E: *D. aff. petiolaris* (Pin cushion); F: *D. species* from Tanami Desert. Photos: A, B, F – R. Nunn; C, E – A. Lowrie; D – T. Handasyde.

Drosera aff. *lanata* from NT (Fig. 3F)

Only found in sandy habitats to the southeast of Darwin, Northern Territory. In the wet season, this species has few hairs covering its leaves in the wet season. By contrast, these same leaves are densely hairy, like those of *D. lanata*, in the dry season.

Drosera aff. *petiolaris* from NT (Fig. 4C)

The N.T. taxon is actually an undescribed species of perennial tropical *Drosera*. It has leaves up to 12 cm long and an inflorescence that has semi-erect pedicels 3-8mm long. By contrast, type *D. petiolaris* from Queensland has leaves up to 6 cm long and an inflorescence with pendulous pedicels 1.5-2.5 mm long.

Drosera aff. *petiolaris* (Pin cushion) (Fig. 4E)

A clumping species bearing very narrow leaves that are both erect and semi-erect within each leafy rosette, appearing pin-cushion like overall.

Drosera aff. *paradoxa* (Orange flower) (Fig. 4A)

Drosera aff. *paradoxa* (orange flower) is distinguished from *D. paradoxa* by its overall smaller habit and metallic orange flowers, as well as its mostly annual habit in the wild. *Drosera* aff. *paradoxa* (orange flower) generally grows in very shallow skeletal soils over sandstone rock which dries out quickly with the onset of the dry season.

Drosera aff. *paradoxa* (Swamp form) (Fig. 4B)

Drosera aff. *paradoxa* (swamp form) is distinguished from *D. paradoxa* by its lack of an erect, woody stem on mature plants, its clumping habit, and preference for saturated, black silty soils in wet season flooded herb fields.

The six seasons of tropical northern Australia

Aboriginal Australians of the Northern Territory of Australia recognise six different seasons based on thousands of years of local knowledge (Parks Australia 2021). The transitions between seasons are marked by subtle variations in the weather, the plants in flower, and the bush foods that are most abundant. The tropical *Drosera* of section *Lasiocephala* have adapted in unique ways to survive this harsh and unforgiving climate.

Kudjewk – Monsoon season

December to March. 24°C - 34°C

This is the true tropical summer season. Thunderstorms, heavy rain and flooding prevail. The heat and humidity cause an explosion of plant life. Speargrass the dominant grass of northern Australia grows to over two metres tall and overall gives a silvery-green appearance to the woodlands.

Bangkerreng – ‘Knock ‘em down’ storm season

April. 23°C - 34°C

By April the rain clouds have dispersed and clear skies prevail. The vast expanses of floodwater recede and streams start to run clear again and the land is marked by violent, windy storms that flatten the speargrass – these are called ‘knock ‘em down’ storms.

Yekke – Cooler (but still humid) season

May to mid-June. 21°C - 33°C

This is a relatively cool time with less humidity and dry winds. Early-morning mists hang low over the plains and to not only to thoroughly soak your bedding when camped out under the stars but also watering the rich and variable flora array of the herb fields each day.

Wurrkeng – Cold weather season

Mid-June to mid-August. 17°C - 32°C

This is the cold time by Tropical Northern Australian standards. Humidity is low, while daytime temperatures are around 30°C and drop to about 17°C at night. Most creeks stop flowing and the floodplains quickly dry out but are refreshed by the dew at night.

Kurrung – Hot dry weather season

Mid-August to mid-October. 23°C - 37°C

The country is hot and dry and dusty as the thunderclouds start to build.

Kunumeleng – Pre-monsoon storm season

Mid-October to late December. 24°C - 37°C

The pre-monsoon season's hot weather becomes increasingly humid. It can last from a few weeks to several months. Thunderstorms build in the afternoons and their showers bring green to the dry land.

Dormancy and survival strategies

The Tropical *Drosera* have developed some remarkable adaptations and clever strategies to maintain the wellbeing and good health of their life-forms. Stormy and cyclonic weather events, droughts, flooding, and fires are regularly encountered over their variable landform habitats each season. In every case each species has successfully evolved its own strategies to out-manoeuvre every one of these natural disasters.

Different Tropical *Drosera* species have evolved to utilise every form of habitat and landform across tropical northern Australia from east to west. From the driest desert areas of inland Australia to the wettest swamps and everywhere in between has been colonised by the tropical *Drosera* species of Australia. It is not uncommon to see examples of hilly landforms with a wet zone at the base of its hill where the soil types are lateritic loam on the summit of the hill, a mixture of sand and laterite soils on the downward scree slopes, and grey silty soils on the flat land immediately below the hill. Commonly in this landscape especially about the Darwin districts, one would commonly find: *Drosera darwinensis* towards the hills' summit; *D. brevicornis* on the scree slopes of the hill; and *D. falconeri* on the flatland in the grey silty soil.

The foliage of many species of tropical *Drosera* is covered by a hairy indumentum. Each hair can be entire or a branching (dendritic) hair. When these hairs are tightly packed together over the foliage, they produce a rather woolly appearance to all of the plant. In full dormancy the rosettes of these species dry back to their central parts to form a tight compact cluster of spent leaf bases that are still covered in the remains of their woolly indumentum. This adaptation strategy is remarkably fire proof against any passing fire event. Tropical northern Australia is commonly subjected to bush fires that are started by electrical storms' lightning strikes.

The very outer parts of the dormant rosette are sacrificed to the flames until the fire quickly passes over them and the central live parts of the plant are safe and protected from the flames. Ad-

ditionally, this sacrificial outer spent leaf strategy produces a secondary cooling effect by dragging cooler air throughout the central parts of the rosette as the fire's heat rapidly rises and exits the burning rosette. The passing fire's fuel load quickly burns out the moment the outer sacrificial leaves have been burnt away.

In tropical northern Australia the seasonal burning of the country stimulates new and rapid grass growth. This method has been practiced seasonally by aboriginal man for eons to provide food to the grazing animals and also to congregate those same animals into the same area where they can be hunted easily for food. Cattlemen of tropical northern Australia within the same country also practice fire farming to promote new grass growth. They call it "green pick". Remarkably grass shoots appear in just a matter of a few short days after a burn.

One of us (A. Lowrie) has witnessed a large *Drosera ordensis* colony near Kununurra, WA, that had a vehicle track cut through the middle of the population. On one side of the track, it had been burnt by a grass fire, on the other side of the track it was unburnt because the track had acted as a fire break. All plants in the population were fully dormant and growing in dry sandy soil. Those plants outer parts on the burn side of the track were singed and blackened but still fully alive and starting to produce new leaf growth as well as producing flowering inflorescences. While those plants on the unburnt trackside were still fully dormant and showing no signs of any new growth. It appears fire or the chemicals from fire, is a natural stimulate for foliage growth.

Species having a woolly indumentum also are quite clever at using their hairy covering as a self-watering system. Utilizing the moisture derived from the early morning dews in the dry season, these species have the ability to harvest water for their needs. They do this by attracting moisture from the foggy morning air. Each hair tip captures a minute bead of moisture which in turn flows downward to join with other minute bead of moisture together accumulating into ever more large droplets that eventually drop to the soil surface as a large droplet and by default water the root zone base of the plant. Some of these woolly species especially those from those rather arid habitats also have many branching hairs along each individual hair within their indumentum. These dendritic hairs increase the area of moisture capture from the foggy air.

New root growth consists of a few water-filled fleshy rather fat parsnip-like roots that penetrate the soil at the start of the plant's new season growth cycle. Later in the season these same few fleshy roots lose their moisture and in turn revert to an extremely strong thin wire-like root that penetrated deep into the ground. Over many seasons these wire-like roots persist and it is not unusual to see when plants are removed from the soil a large number of these wire-like roots attached at the base of the plant. Together these roots anchor the plants to the soil strongly where they hold fast to the soil during any flooding event.

A different dormancy strategy, resting below the soil surface has been adopted by two species: *Drosera falconeri* and *Drosera kenneallyi*. Both of these species retreat to their compact bulb-like fleshy leaf bases. This structure can be likened to that same structure of *Dionaea muscipula* (Venus flytrap), simply a bulb-like accumulation of compact fleshy leaf bases that it uses as a dormancy adaptation.

Both of these species favour monsoon period flooded grey silty soils generally where grey magnetic ant mounds are often found. The bulb-like structure of these plants is positioned just below the soil surface, above which the leafy rosettes lay prostrate hard pressed to the soil surface. At the end of its leafy growth above the soil surface, the leaves are deciduous and the plants retreat to their bulb-like bases below the soil surface. At this time these same saturated soils have dried out, hardened to the point where the soil has the consistency of an adobe brick. It is here cocooned in its concrete-like surrounds that it will spend its entire dormancy during the hot dry season and safe from any bushfires.

Growth

The dormancy of tropical *Drosera* species is broken with the first rain showers during the build-up to the wet season. The annual species germinate quickly from seed and grow rapidly as a result of the available moisture and also the ability to capture and digest prey in the early stages of growth. Perennial *Drosera* develop new leaves quickly from the centre of the bulb-like structure, and in some species, the inflorescence is also produced at this time. From the first moment that moisture is available, the race is on to renew vegetative growth, build up reserves, flower and disperse seed. This urgency is necessary, as in some years, the wet season is short and sudden droughts occurs. At the beginning of the growing season new roots are produced. These roots are white, thick and fleshy, and hold a considerable amount of moisture. At the end of the season, these same roots become thin and fibrous. They then lack the fleshy structure needed to hold moisture and are simply employed to anchor the plant to the soil. It is not known whether these fibrous roots are still capable of absorbing moisture.

Flowering

Each inflorescence generally presents only one open flower at a time. The flower lasts one day whether pollinated or not. All species have inflorescences that support many flower buds. Additionally, each species produces more than one inflorescence. *Drosera falconeri* generally has an inflorescence supporting about 12 flowers, so the flowering period is short. *D. fulva* commonly produces three scapes up to 45 cm tall, each supporting inflorescences holding 50 plus flowers. The large number of flowers produced means that *D. fulva* can be found in flower from February through to May.

After the first monsoon rain, most of the tropical *Drosera* species produce their juvenile or somewhat advanced flowering inflorescences at the same time that they are also renewing their new season's leafy growth. Two species however, *D. falconeri* and *D. kenneallyi*, employ a totally different flowering regime. During the build-up (the pre-monsoon season), when the soil is still reasonably dry, both of these species start to flower well before any meaningful foliage is produced. They flower, are pollinated, and when spent get pushed over to the soil surface by the production of their new season leafy rosettes that have developed well after their flowering events have ended. The seed pods still attached to their spent inflorescences are still held in their seed capsules but are now covered by the leaves pressing them to the soil surface. Here they will remain until they are released when all leafy growth has dried out.

Drosera caduca is a strange species as it is only at its insect capture stage at the beginning of its seasonally new first growth and then only for a short period of time. After its carnivorous growth stage, it reverts to its non-glandular long grass like lamina. At this same time, it also produces its flowers. Later in the season all of this species foliage and all parts above the soil become deciduous and the plants retreat to a bulb-like structure made up of the fleshy bases of the foliage into their sandy soil habitat. Just the tips of these structures are at the soil surface where they remain while they are dormant. This species asexually reproduces itself and this results in large clumps growing in compact clusters. At the time of dormancy those plants derived from the mother plant have mostly detached from each other.

The colours of the flowers of the perennial tropical *Drosera* (Figs. 5A to 5D) vary from white through to dark pink and one still unnamed taxon has orange flowers. It is not uncommon for a particular species to have white and pink coloured flower forms. Morphologically, there are very few good floral characteristics that distinguish one species from the next. One exception is *D. brevicornis*, which has a strange, large, hook-like projection above each pair of anthers on all five stamens. Pedicel length and its position (erect, semi-erect, or pendulous) in fruit on the inflorescence can be used as a

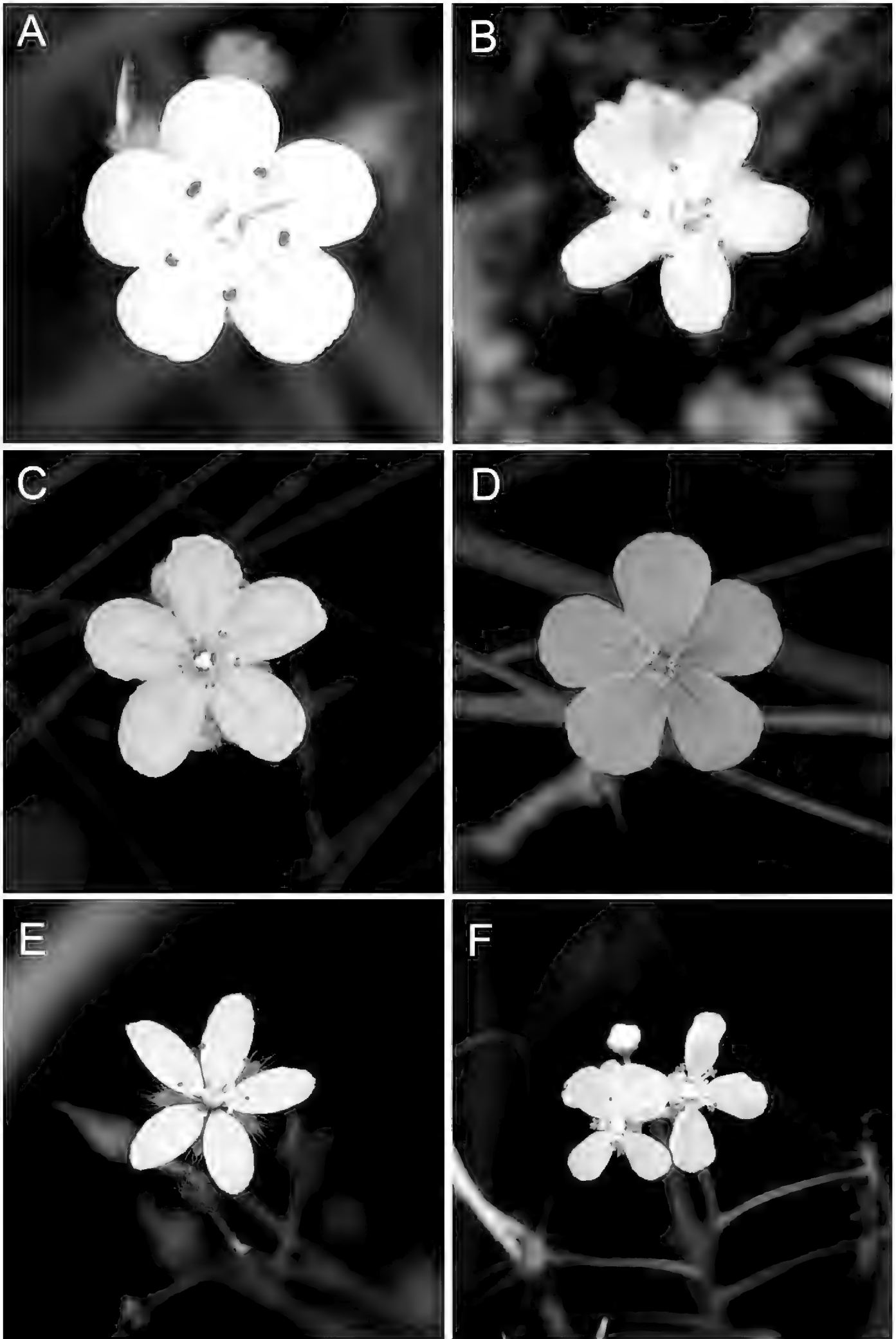


Figure 5: A: *Drosera lanata*; B: *D. petiolaris*; C: *D. brevicornis*; D: *D. aff. paradoxa* 'Orange flower'; E: *D. banksii*; F: *D. subtilis*. Photos: R. Nunn.

characteristic to help distinguish one species from the next. Conversely, the hairy indumentum of the inflorescence appears to be similar amongst most named species in this complex. It should also be noted that this hairy covering is commonly mixed with dendritic hairs. *Drosera broomensis* is the only odd one out, with its glabrous scape and inflorescence. The flowers of the two annual species (Figs. 5E & 5F) are inconspicuous and white but have useful characters to easily identify each species.

Acknowledgements: The authors would like to thank Greg Bourke, Alastair Robinson, Paulo Baleeiro and Tricia Handasyde, for providing images without which this work would not be complete. Jan Schlauer and Andreas Fleischmann for reviewing the manuscript and providing many useful observations and comments.

References

- Baleeiro P.C., Jobson, R.W., and Barrett, R.L. 2020. *Drosera stipularis*, a new species for the *D. petiolaris* complex from Cape York Peninsula, Queensland. *Telopea* 23: 35-40.
- Bourke, G., and Nunn, R. 2012. Australian Carnivorous Plants. Redfern Natural History Productions Ltd.: Dorset, UK.
- de Candolle, A.P. 1824. *Prodromus systematis naturalis regni vegetabilis* 1. Sumptibus Sociorum Treuttel et Wurtz: Paris.
- Diels, F.L.E. 1906. Droseraceae. In: Engler, H.G.A. (ed.). *Das Pflanzenreich* 4: 1-136.
- Fleischmann, A., Cross, A.T., Gibson, R., Gonella, P.M., and Dixon, K.W. 2018. Systematics and evolution of Droseraceae. In: Ellison, A.M., and Adamec, L. (eds.) *Carnivorous Plants: Physiology, ecology, and evolution*. Oxford University Press: Oxford. pp. 45-57.
- Kondo, K. 1984. Three new species of *Drosera* L. from Australia. *Boletim da Sociedade Broteriana* 57: 51-60.
- Lowrie, A. 1994. *Drosera ordensis* (Droseraceae), a new tropical species of carnivorous plant from northern Australia. *Nuytsia* 9: 363-367.
- Lowrie, A. 1996a. *Drosera kenneallyi* (Droseraceae), a new tropical species of carnivorous plant from the Kimberley, northern Western Australia. *Nuytsia* 10: 419-423.
- Lowrie, A. 1996b. New species in *Drosera* section *Lasiocephala* (Droseraceae) from tropical northern Australia. *Nuytsia* 11: 55-69.
- Lowrie, A. 1997. *Drosera paradoxa* (Droseraceae), a new species from northern Australia. *Nuytsia* 11: 347-351.
- Lowrie, A. 1998. *Carnivorous Plants of Australia*, Volume 3. University of Western Australia Press.
- Lowrie, A. 2014. *Carnivorous Plants of Australia*, Magnum Opus, Volume 2. Redfern Natural History Productions Ltd.: Dorset, UK.
- Lowrie, A., Robinson, A., Nunn, R., Rice, B., Bourke, G., Gibson, R., McPherson, S., and Fleischmann, A. 2017. *Drosera of the World*, Volume 2: Oceania, Asia, Europe, North America. Redfern Natural History Productions Ltd.
- Marchant, N.G., and George, A.S. 1982. *Drosera*. In: George, A.S. (ed.). *Flora of Australia* 8. Canberra. pp. 9-46.
- Parks Australia. 2021. Six Seasons. Australian Government, Kakadu National Park. <https://parksaustralia.gov.au/kakadu/discover/nature/seasons/>
- Planchon, J.E. 1848. Sur la famille des Droséracées. *Annales des sciences naturelles. Botanique sér.* 2 Sér. 3: 79-98, 185-207, 285-309.
- Seine, R., and Barthlott, W. 1994. Some proposals on the infrageneric classification of *Drosera* L. *Taxon* 43: 583-589.

CARNIVOROUS PLANTS OF AUSTRALIA’S TOP END

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Keywords: Top End, Northern Territory, Australia, *Utricularia*, *Drosera*, *Byblis*.

Abstract: The Top End of the Northern Territory in Australia is a centre of diversity for carnivorous plants. In May 2021, Boaz Ng embarked on a 10-day expedition to document these plants in the wild and photographed over 45 taxa across 3 genera. This field report recounts his experience and focuses on a selection of species that represent adaptations towards the wet and dry seasonal dichotomy of the region.

Acknowledgement of Country: The author would like to acknowledge the Traditional Owners of the land on which this expedition took place, including the Larrakia, Werat, Koongurrukun, Wajray, Marranunggu, Marrathiel, Wulna, Limilngan, Ngomburr, Gun-djeihmi, Warramal, Gaagudju, Bukurnidja, Konbudj, Mbukarla, Amarak, Wagiman, Mangarrayi, Yangman and Jawoyn Peoples, among others. Their continued custodianship of the land and protection of its biodiversity is especially relevant to this article.

A blast of heat hit my face as if the doors of a furnace were opened. Instantly, beads of sweat materialized and made me wonder how I was going to possibly make it through 10 entire days of botanizing in the oppressive tropical climate. After a restless night spent dreaming about the carnivorous plants of the Top End, an agitated sensation comprising half excitement and half delirium began to fill the car, quickly building thicker than the humidity in the air.

The “Top End”, an Australianism for the tropical regions of the Northern Territory, is known for its two strikingly dichotomous seasons – the “wet” with daily monsoonal downpours and the “dry” in which not a single drop of rain falls. This is an oversimplification, with the Indigenous peoples of the region recognizing up to six distinct seasons. Our expedition was timed for the start of *Yekke*¹, after the floodwaters recede, gambling that the La Niña conditions would prolong the wetness and that we would catch the narrow period when the greatest number of carnivorous plants can be seen.

My eyes glanced nervously for signs of water as we arrived at the Howard Sand Plains, a habitat formed by a thick deposit of sand in the floodplain the Howard River east of Darwin. With the onset of the monsoon, the river swells and inundates the sand sheets. When the rains cease, the water rapidly drains to sea and exposes the substrate to air once again. It is during this brief transitional period between the wet and dry that the bladderworts, of which there are at least 26 species in the sand plains alone, come to bloom. In the trail, deeply eroded ravines hinted of the now vanished floodwaters and I anxiously approached the steadily browning grasses flanking the track, hoping that I hadn’t arrived too late.

Suddenly I found myself surrounded in a beautiful field of *Utricularia leptoplectra*, with hundreds of purple lobed flowers swaying gently in the breeze. Glistening specimens of *Drosera aquatica* (Fig. 1) glowed like jewels amongst the damp shadows as they caught the morning sun with their spidery arms. As its name suggests, the species is adapted to float in floodwaters and is even known to complete its life cycle without ever touching the ground. The water table was still high enough to flow just under the sandy surface and seeped out to form shallow channels wherever old trails had

¹ A season of the Kakadu region in the Kunwunjku and Kundjeyhmi languages (among others). Each language group has their own names and interpretations of the various seasons.



Figure 1: *Drosera aquatica*.

been dug. At their edges grew *U. circumvoluta*, with a twining peduncle that spirals around sedges for support, and inside the pools I even found *U. adamsii*, a recently described floating species that occupies shallow wetlands. I was amazed at the diversity in both plants and environmental niches, having spotted 15 species in a trail just 500 meters in length.

The following day, we embarked on our road trip around the Top End and drove out of the city towards Berry Springs. Along the way, I had identified several sites where I hoped to find the iconic *D. falconeri* (Fig. 2; Front Cover). Renowned for its disc-like laminae that are reminiscent of the Venus flytrap, the species has captured the imaginations of enthusiasts worldwide and no pilgrimage to the Territory would be complete without sighting the plant.

I soon discovered out that finding it was going to be more challenging than I thought. The species is mainly known from the floodplains of major rivers south west of Darwin, occupying sandy-silt alluvial deposits that lie beneath grasses so thick and tall that you can barely see through them. Luckily by this time, they were almost desiccated and snapped without resistance. I tried my best to ignore the irritation from the increasing number of sharp seeds lodged in my socks as I traced the banks of the dried three-meter-deep ravine. Noticing that the soil was silty and grey, I kicked down some grasses to expose what I had come to find.

Like other members of the section *Lasiocephala*, *D. falconeri* is well adapted for the climatic dichotomy of the region. With the onset of the dry season, it retreats underground as a resting corm, using the now rock-hard substrate as insulation against heat. This habit means that the species is amongst the first in the complex to enter dormancy, dropping its leaves as soon as the rains stop. Unfortunately, the plants were already well past their dewy prime by the time I had arrived. Whilst an observation of dormant plants contributes valuable field knowledge, I was admittedly disappointed, and I trudged back to the car rather scratched up from the grasses I had to push through.



Figure 2: *Drosera falconeri*.

We awoke before sunrise in preparation for a massive day in the Litchfield National Park. The region is known for its gigantic waterfalls that plunge off cliffs into the plains below and it was in the watercourses of the sandstone escarpment that we focused our search. Timing the exploration of these creeks is always risky – arrive too early and they're still raging torrents too forceful for plants; too late and they've already dried out, the waters from the elevated plateau disappearing especially fast.

The major stop for the day was a wide-banked cascade flowing off the escarpment that I was hopeful would provide just the right balance in size to support carnivorous plants. I soon found out that getting there would be no easy feat. The oppressive humidity and searing heat in the exposed plateau turned what would otherwise be a gently undulating stroll into a horrible slog. After what seemed like hours of almost collapsing from heat stroke, the caressing sound of flowing water gave me a burst of energy required to make the final stretch. Plunging into a rock pool fully clothed, I was instantly rejuvenated with the sight of *D. burmannii* at the water's edge.

In the fast-flowing waters, I saw the leaves of *U. fulva* growing directly in the current. However only a few scattered blooms were present – far from the large flowering events I was hoping to photograph. Further downstream, amazing specimens of *Byblis liniflora* (Fig. 3) grew directly in the rock face. Just a month or two ago, the site would have been covered by the stream and in a few weeks, the waters will stop supplying the fractures the plants occupy. For this species, seed is an important mechanism for surviving both the wet and dry seasons, wedging themselves tightly within the tiniest cracks until the water level is just right to spring forth a new generation of plants.

After the Litchfield National Park, we circuited back to greater Darwin where we spent the day surveying a few dams and lagoons. Unlike the ephemeral sand plain wetlands, these permanent



Figure 3: *Byblis liniflora*.

bodies of water can support obligate aquatic plants such as the suspended aquatic bladderworts. The deep-water habitats are also home to the deadly saltwater crocodile...

We arrived at a lagoon in the outer suburbs to have lunch. At the picnic area, a dilapidated hand-made sign warned of crocodiles, which are known to sometimes slip through the caged traps set to relocate them. My heart began to race as I spied the water's edge, nervously keeping one eye out for crocs. I was enticed by gigantic strands of *U. muelleri* (Fig. 4) floating in the clear waters, each stem reaching over a meter long with hundreds of bladder traps arranged on bushy whorls of leaves. With a shot of adrenaline, I approached the water for just a few seconds to take images before retreating to the safety of higher ground.

While a permanently wet habitat provides a stress-free lifestyle for the suspended aquatic bladderworts, it also presents a problem when the plants are trying to reproduce sexually. Whereas the terrestrial bladderworts can simply produce a flower stem anchored off the ground, the floating species must somehow elevate their blooms above the surface of the water and keep them stable enough for insects to land on them. *Utricularia muelleri* has evolved an elegant solution and sends up a long stem with a star-like structure at the end. When this structure reaches the surface, it inflates into a gas-filled float, out of which emerges the inflorescence. This float ensures that the blooms are held stably above the surface of the water, allowing for efficient pollination.

By now we were nearly halfway through our journey, with the next three days to be spent in the iconic Kakadu National Park. Situated at the western edge of the Arnhem Plateau, the region is characterized by its dramatic cliffs, vast wetlands and ancient cultural heritage. As we drove into the area, we noticed smoke emanating from the highway and even saw flames lapping by the roads edge!

The wet season produces an enormous number of tall grasses that completely carpet the landscape. When the rain stops, the grasses dehydrate into thickets of extremely flammable tinder. Wide-



Figure 4: *Utricularia muelleri*.

spread fires grip the countryside during the dry season, so low-intensity controlled burns are utilized to create strategic firebreaks and rejuvenate the land. To me, the fires enhanced the spirituality of the sacred landscape. Seeing the Burrungkuy outcrop, an ancient meeting place with rock art dating back more than 20,000 years, glow an incandescent red as the evening sun filtered through the smoke was surely one of the most reflective experiences of my life. I was also left to wonder how the sundews growing in the area would cope with the fires.

As we left Kakadu, I inspected a section of forest that was burned just a few days before. Scattered in the undergrowth were rosettes of *D. dilatatopetiolaris* (Fig. 5), which had begun to enter its dormant phase by reducing the size of their leaves and increasing the amount of trichomes produced on their stems. This results in a densely packed bud of white hairs, which gives the complex the common name of the “woolly sundews”. In patches which were burnt, I noticed the singed remains of old rosettes, but the hairy resting bud seemed to be able resist the flames. I find it amazing that these tough plants had to not only survive the annual drought, but also the fire ubiquitous in their habitat.

Our journey continued south towards Katherine, which is known for its majestic gorges and plunging waterfalls that fill the landscape of the Nitmiluk National Park. This region is placed at the transition zone between the wet tropics and arid interior of the Territory. Being situated inland, the area was well and truly in the grips of the dry season and I was worried that I had arrived too late.

We started the hike up the escarpment early in the day and was surprised by the decidedly bitter morning air. I was on the lookout out for small ephemeral streamlets and wet skeletal soils over the sandstone bedrock, a niche that is almost always associated with carnivorous plants in Australia. While the stained rocks and eroded patterns hinted of their presence, they had long been desiccated by the heat that was really beginning to build up. The main river itself was too large and forceful to support populations of bladderworts so I turned my attention towards a dry creek bed and followed it upstream.



Figure 5: *Drosera dilatatopetiolaris*.



Figure 6: *Utricularia fulva*.

As I followed the creek, the substrate started to moisten and soon a trickle of water started flowing. I was relieved to find that the sides of the rivulets were painted orange from the myriads of *U. fulva* (Fig. 6) that were blooming en masse. The species is notable for its attractive tawny and brick-red petals, which are colored just like the sandstone on which it grows. In contrast to the flowerless leaves I had seen in the Litchfield, which were submerged under a fast-flowing stream, the plants here were now located on the steadily receding sandbanks. The late blooming period allows the species to recolonize the site from seed, rather than having it washed away by the raging torrent present at the height of the wet season.

On the final day of the journey, I returned to the Howard Sand Plains for a full day of exploration. The water table had dropped significantly in just 10 days, and the sites I had explored before were visibly drier. The floating stems of *U. adamsii* had perished and were replaced by the flowers of *U. gibba*, a species that is induced to bloom when its aquatic stolons are stranded in drying mud. There was still one particularly rare species that I wanted to find. With the relentless sun directly above me, I trekked along an old trail deep into the bush, blindly hoping that the site would somehow still be wet enough. After an hour, I came across some rather gothic looking magnetic termite mounds, a biological beacon signalling that I had arrived at the right place in the floodplain.

Delighted that the ground was soaked, I dropped my backpack and scoured the area for the elusive plant. The five-lobed *U. holtzei*, pin-sized *U. quinquedentata*, and showy *U. odorata* were everywhere but these weren't the bladderwort that I was trying to find. After an hour spent searching a plot of around twenty square meters, the oppressive climate overcame me. As I bent over to pick up my bag, I saw an alien bloom with two long devil-like prongs and a gaping beak staring straight at me! Despite its somewhat generic habitat, *U. dunstaniae* (Fig. 7) is notoriously difficult to observe, perhaps due to a very specific niche or narrow flowering period. The water level in the




Figure 7: *Utricularia dunstaniae*.

area had receded such that only a percolating pool of water a centimeter deep remained, and it was in this finely balanced microhabitat that I found the elusive species.

Behind the drops of sweat, a slight grin crept across my face as I realized my gamble in timing had paid off. Incredible luck is needed when botanizing in the Top End, and you just have to find yourself in the right place at the right time. Had I visited this site on the first day of the expedition, it would have been too flooded to induce flowering, and in another week this puddle will disappear into thin air. Likewise, the *Utricularia* of the sand plains will soon perish, persisting only as seed that awaits the monsoon rains in an endless cycle of wet and dry.


Acknowledgements: The author thanks Peter Eggenhuizen and Wey Yao Wong for their companionship throughout the trip; Richard Nunn, Andrew Broome, and Thilo Krueger for their insight and discussions of the field; and the broader carnivorous plant community, whose support motivates me to find these amazing plants.




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WAUKAUYENG TIPU AND UCHII RIVER UPLANDS EXPEDITION 2019 – REPORT WITH PHOTOGRAPHIC OBSERVATIONS OF CARNIVOROUS FLORA OF THE PARUIMA TERRITORY IN THE CUYUNI-MAZARUNI REGION OF GUYANA

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Keywords: Guiana Highlands, Waukauyengtipu, tepui, Guiana Shield, Pantepui, carnivorous plants, Uchii River, Gran Sabana.

Abstract: An expedition to the territory of Paruima and its vicinities was organized between January 3-12, 2019. The objective was to visit the areas belonging to the Guyanese outreach of the Gran Sabana (Eng. “Great Savanna”) in the vicinity of the Uchii River (aka Utshi River) near the Venezuelan border and to reach the underexplored summit of the Waukauyengtipu plateau. The aim of the expedition was to make botanical observations with the main focus of reviewing local carnivorous plant species occurrences, as well as to assess the feasibility of the area for carrying out future scientific research and implementing small-scale eco-tourism activities in the territories belonging to the Pemón Arekuna community of Paruima. The expedition resulted in making interesting observations regarding the unique flora of the Guiana Highlands found around the areas of the Uchii River uplands in the Gran Sabana and on Waukauyengtipu.

Introduction

An expedition to the territory of Paruima and its vicinity was organized between January 3-12, 2019. The objective was to visit the areas belonging to the Guyanese outreach of the Gran Sabana (Eng. “Great Savanna”) in the vicinity of the Uchii River (aka Utshi River) and Uchii Falls (aka Utshi Falls) near the Venezuelan borderline and to reach the underexplored summit of the Waukauyengtipu plateau.

The aim of the expedition was to make photographic botanical observations with the main focus being reviewing local carnivorous plant species occurrences, as well as to assess the feasibility of the area for carrying out possible future scientific research and implementing small-scale eco-tourism activities in the territories belonging to the Pemón Arekuna community of Paruima.

The areas surrounding Paruima offer access to large expanses of premontane and montane primary tropical forest. In addition, shrub savannas and savanna/forest mosaic areas are in close proximity to Uchii Falls and the Venezuelan borderline marked by the so-called Schomburgk Line. Paruima and its vicinity also offer a number of habitats associated with the summits of the tepuis (Kelloff *et al.* 2011, pp. 25-29), or the Pantepui biogeographical region in general, most notably on the summit areas of Waukauyengtipu. Waukauyengtipu is a table mountain situated close to the western border of Guyana marked by the course of the Venamo (aka Wenamu) River (Anderson 2003) and the northwestern end of the so-called Schomburgk Line, and is one of the highest mountains in Guyana (Kelloff *et al.* 2011, pp. 44-46).

The expedition was co-organized independently by the author of this paper in collaboration with Darrell Carpenay, a Guyanese photographer. The team consisted of four people accompanied by members of the Indigenous Pemón Arekuna community of Paruima:

- Mateusz Wrazidlo – co-organizer and lead coordinator,
- Darrell Carpenay – co-organizer and photographer,

- Orson Hinds – logistics coordinator and expedition chef,
- Izabela Stachowicz – biologist and scientific consultant,
- Alex Smith – guide for Uchii Falls and Waukauyengtipu,
- Calio Elliman – guide for Waukauyengtipu and local plant expert,
- Charlie Elliman – assistant guide for Waukauyengtipu.

The organizational process for visiting the area was coordinated by Lee Williams (Toshao of Paruima) and Lennox Percy (vice-Toshao of Paruima).

Materials and methods

A survey in Region 7, Cuyuni-Mazaruni in Guyana was carried out during an expedition organized in January 2019. The methodology was based on making in-situ observations, supplemented by preparing photographic documentation of a number of carnivorous plant habitats encountered along the trekking path. In total, 5 perimeters were visited and documented, as listed in Table 1. Plant photographs were made using a digital camera equipped with a zoom lens (Olympus OM-D E-M5 mk.II + Panasonic Leica DG Vario-Elmarit 12-60 mm f/2.8-4 ASPH). Taxonomic identification of the documented specimens was based on a morphological analysis according to Taylor

Table 1. Observation perimeters with recorded carnivorous plant occurrences.				
No.	Observation perimeter name	Approx. coordinates	Approx. elevation [m a.s.l.]	Carnivorous plant species observed
1	Savannas around Uchii Falls	5°41'11.9"N 61°06'44.4"W	860-900	<i>Catopsis berteroniana</i> <i>Drosera kaieteurensis</i> <i>Utricularia amethystina</i> <i>Utricularia longeciliata</i> <i>Utricularia trichophylla</i> <i>Utricularia pubescens</i> <i>Utricularia hispida</i>
2	Mosaic forest & savannas leading to Venezuelan border	5°40'21.5"N 61°07'12.1"W	900-1000	<i>Brocchinia reducta</i> <i>Catopsis berteroniana</i> <i>Drosera felix</i> <i>Utricularia hispida</i> <i>Utricularia jamesoniana</i> <i>Utricularia pusilla</i> <i>Utricularia subulata</i>
3	Trail from Ararata Camp to Waukauyengtipu wall	5°49'37.6"N 61°08'40.0"W – 5°49'41.4"N 61°11'40.8"W	890-1350	<i>Utricularia</i> cf. <i>alpina</i> <i>Utricularia jamesoniana</i>
4	Waukauyengtipu summit – vicinities of Camp Misery	5°49'48.1"N 61°12'32.7"W	1430-1510	<i>Utricularia</i> cf. <i>alpina</i> <i>Utricularia jamesoniana</i>
5	Waukauyengtipu summit – Stegolepis wetland	5°49'56.9"N 61°14'05.9"W	1450	<i>Brocchinia reducta</i> <i>Heliamphora heterodoxa</i> <i>Drosera roraimae</i> <i>Drosera kaieteurensis</i>

(1989) for *Lentibulariaceae*, McPherson *et al.* (2011) for *Sarraceniaceae*, Robinson *et al.* (2017) for *Droseraceae*, and Holst (1997) for *Bromeliaceae*. Elevation data and coordinates were collected using a GPS device (Garmin Oregon 750t).

The locations chosen for the undertaking were the Uchii River uplands in the Guyanese part of the Gran Sabana and the plateau of Waukauyengtipu, which until this day remains one of the underexplored areas of the Guiana Highlands (Huber 1995). The Uchii River uplands were studied botanically in the past, most notably during three expeditions being parts of the Smithsonian Biological Diversity of the Guiana Shield Program. The first expedition was led by Tim McDowell and took place in May 1990 (Hollowell *et al.* 2004). The other ones were led by H. David Clarke, between January-February 1996 (Kelloff *et al.* 2011, pp. 25-29) and June-July 1997 (Kelloff *et al.* 2011, pp. 44-46). H. David Clarke's 1997 expedition also includes the only recorded visit to the Waukauyengtipu plateau prior to the expedition hereby reported.

Fieldwork report

Part 1 – Uchii River Uplands

Thursday, January 3

The team assembled at the Ogle Airport in Georgetown, Guyana at 7:00 AM and boarded a charter plane (BN-2 Islander reg. no. 8R-GHM) operated by Trans Guyana Airways at 1:00 PM, following a short delay dictated by unfavorable weather conditions. The plane arrived in Paruima shortly before 2:30 PM. The team members were accommodated in a local health post to prepare for the upcoming trekking activities. A meeting was organized with the Community Council. The residents of the village were presented with a short presentation about the plans for the expedition. Consultations were carried out regarding the itinerary for the following days. Guides were introduced to the team.

Friday, January 4

The group left Paruima shortly before 7:00 AM and advanced southwards after crossing the Kamarang River, following a forested trail leading to Uchii Falls accompanied by a local guide, Alex Smith. The trail was muddy and steep which eventually contributed to a much-delayed arrival at the final destination. The team left the rainforest and entered the Gran Sabana areas dominated by a notable abundance of *Pteridium* sp. ferns at approx. 4:15 PM. Continuing their walk towards the Uchii Falls campsite, at approx. 4:35 PM the group arrived at the bank of the Uchii River, which required traversing. Due to high water levels and a strong current, crossing the river proved to be much more time consuming and hazardous than expected, forcing a decision to use a nearby Pemón household as an emergency campsite for the night, instead of proceeding to the initially planned campsite by the Uchii Falls cliffside.

First carnivorous plant populations were observed:

- *Utricularia trichophylla* – found in a shallow stream right after entering the savanna, shortly after leaving the forest (Fig. 1A, 1B)
- *Utricularia pubescens* – nearby *U. trichophylla* populations described above (Fig. 1C)
- *Utricularia hispida* – as above (Fig. 1I)
- *Drosera kaieteurensis* – on the banks of the Uchii River on exposed peat patches
- *Catopsis berteroniana* – present in large numbers, growing epiphytically on shrubs and trees around the banks of the Uchii River (Fig. 1F)



Figure 1: Carnivorous plants observed around the Uchii Falls savannas (observation perimeter No. 1): A) *Utricularia trichophylla* – inflorescence; B) *Utricularia trichophylla* colony in a stream; C) *Utricularia pubescens*; D) *Drosera kaieurensis*; E) *Utricularia amethystina*; F) *Catopsis berteroniana*; G) *Utricularia longeciliata*; H) *Utricularia longeciliata* – flowerstalk; I) *Utricularia hispida*.

Saturday, January 5

Saturday was spent mostly on botanical exploration of the Uchii Falls areas and savanna/forest mosaic vegetation areas extending towards the Venezuelan border. The camp was moved and set up nearby the Uchii Falls cliffside at approx. 10:00 AM. Further carnivorous plant observations were made on the bank of the Uchii River, right behind the Uchii Falls cliff zone:

- *Utricularia pubescens*
- *Utricularia amethystina* (Fig. 1E)
- *Utricularia longeciliata* (Fig. 1G, 1H)
- *Drosera kaieteurensis* (Fig. 1D)

Carnivorous plant occurrences observed around the Uchii Falls savannas (observation perimeter No. 1) are presented in Figure 1.

Walking towards the Venezuelan border other carnivorous plant species were found in a patch of mid-elevation loose forest vegetation typical to the Gran Sabana areas (at the elevation of approx. 900-1000 m a.s.l.) with a notable presence of *Stegolepis angustata*, *Saxofrid-ericia regalis*, *Clusia* sp., and myrmecophytic members of the *Melastomataceae* family. These species include:

- *Utricularia jamesoniana* (Fig. 2C)
- *Utricularia pusilla* (Fig. 2A, 2B)
- *Brocchinia reducta* (Fig. 2D)
- *Catopsis berteroniana* (Fig. 2H)

The team continued to walk until they reached the last settlement on the Guyanese side of the border in an open, sandy savanna area approx. 1 km from the Venezuelan border. The place turned out to be a good lookout spot, providing an unobstructed view over the Ilu-Tramen Massif and the Venezuelan part of the Gran Sabana (Fig. 3) leading towards the Pemón Arekuna village of Wuarapata.

The vegetation consisted mostly of open grassland dominated by *Axonopus anceps* with patches of various shrubs. Carnivorous plants found by the end of the trail included:

- *Drosera felix* – present on the trail in moist, sandy areas
- *Utricularia subulata* – in shallow pools alongside the trail
- *Utricularia hispida* – frequent yet scattered occurrences among clumps of *Axonopus anceps*
- *Catopsis berteroniana* – growing abundantly on short trees and shrubs (Fig. 2G) visible all around the perimeter of observation

Walking back to the Uchii Falls campsite the weather cleared up looking northwest, providing a view over the southeastern cliffs of the Waukauyengtípu massif. Campsite was reached at approx. 3:30 PM.

Carnivorous plant occurrences observed around the mosaic forest and savannas leading to the Venezuelan border (observation perimeter No. 2) are presented in Figure 2.

Sunday, January 6

Sunday was dedicated entirely to a return trekking to Paruima. The team cleaned up and left the campsite at approx. 7:00 AM and started walking north towards the village. At 8:15 AM, a junction of Uchii and Kamarang rivers was reached and crossed using an improvised raft. The group took a detour to reach the Uchii Falls gorge to see the waterfall from below, arriving under the cliffs at approx. 9:20 AM (Fig. 4). Due to difficult, muddy conditions on the trail combined with the overall fatigue of the team members, the return trek finished in Paruima shortly after 6:00 PM.

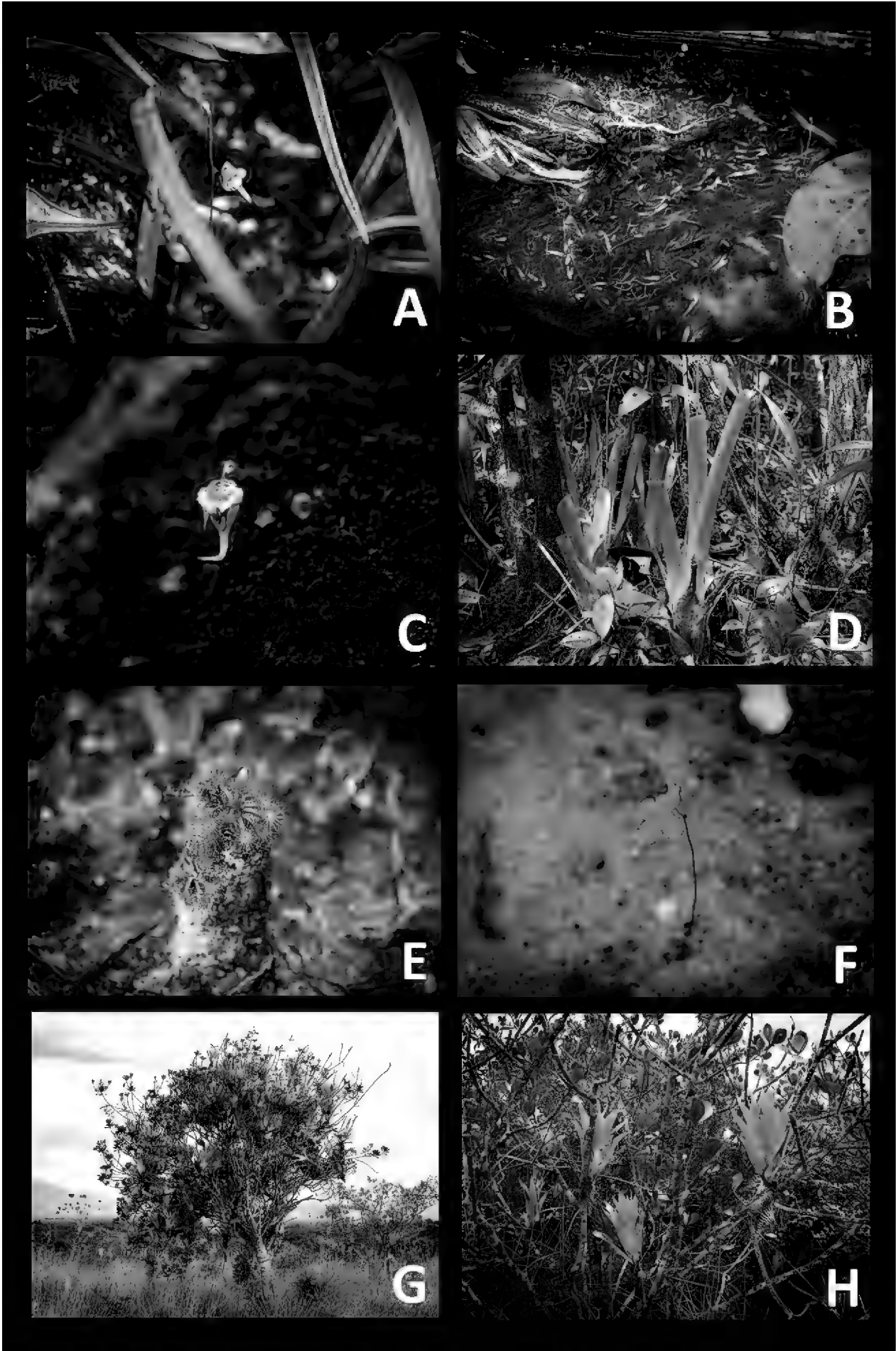


Figure 2: Carnivorous plants observed around the mosaic forest and savannas leading to the Venezuelan border (observation perimeter No. 2): A) *Utricularia pusilla* – inflorescence; B) *Utricularia pusilla* colony in a stream; C) *Utricularia jamesoniana*; D) *Brocchinia reducta*; E) *Drosera felix*; F) *Utricularia subulata*; G) Tree overgrown by a colony of *Catopsis berteroniana*; H) *Catopsis berteroniana*.



Figure 3: Tramen-tepui and Ilu-tepui seen from the Guyanese side of the Gran Sabana, 5°40'12.9"N 61°07'41.9"W, elev. ca. 995 m a.s.l.



Figure 4: Uchii Falls seen from the bottom of the Uchii River gorge. 5°41'25.5"N 61°06'36.6"W, elev. ca. 620 m a.s.l.

Part 2 – Waukauyengtipu

Monday, January 7

After meeting the guides to accompany the group during the second part of the expedition – the ascent of Waukauyengtipu – a boat was used to cross the Kamarang River at approx. 10:00 AM. The trail led westwards and ended with a lower cloud forest right before turning into a patch of sandy plain with scrub vegetation (elevation approx. 890 m a.s.l.) where the campsite was located. The forest was characterized by an abundance of epiphytes, such as *Philodendron insigne* plants, members of the *Bromeliaceae* family, *Rapataea steyermarkii*, and many epiphytic orchids. The first occurrence of a carnivorous plant was confirmed, namely *Utricularia jamesoniana* (Fig. 8A). The campsite, called “Ararata” in the Pemón Arekuna language (meaning exactly “a forest which is not dense” according to Calio Elliman) was reached shortly after 3:00 PM. The campsite provided a clear view over the eastern wall of Waukauyengtipu (Fig. 5).

Tuesday, January 8

The group left the campsite before 8:00 AM and advanced westwards towards the eastern wall of Waukauyengtipu. On the way to the wall, at the elevation of approx. 1330 m a.s.l. populations of *Utricularia* cf. *alpina* were found growing epiphytically on the tree branches. The base of the tepui wall was reached shortly before 1:00 PM, the area right underneath the wall was a wet, boggy ridge. Reaching the summit of the plateau required climbing a vertical, densely vegetated part of the trail



Figure 5: Wall of Waukauyengtipu seen from the Ararata Camp. 5°49'37.6"N 61°08'40.0"W, elev. ca. 890 m a.s.l.



Figure 6: Highlights from the trail to Waukauyengtipu summit: A) Alex Smith crossing over the vertical part of the “Clarke’s Pass”, approx. elev. 1430 m a.s.l.; B) Cloud forest on the summit of Waukauyengtipu.

(dubbed “Clarke’s Pass”, referring to the descriptions left by the leader of the 1997 Smithsonian Expedition) using only exposed roots (Fig. 6A). The summit of Waukauyengtipu turned out to be covered by a dense, damp Pantepui cloud forest (Fig. 6B). After walking approx. 1.8 km across the summit plateau, the campsite, called “Camp Misery” due to its cold and wet surroundings at the elevation of approx. 1510 m a.s.l., was reached at approx. 3:30 PM.

Wednesday, January 9

On Wednesday the group split into two smaller teams – Darrell, Orson and Izabela retreated back to camp Ararata, while Mateusz and Calio left the campsite before 7:00 AM and continued westwards, followed by Alex and Charlie, with the objective to push towards one of the scattered open peat wetland vegetation areas on the summit of Waukauyengtipu. The summit turned out to be a densely vegetated, swampy area which was exceptionally laborious to penetrate, and characterized by an abundance of plants typical to the Pantepui cloud forest vegetation, such as *Brocchinia tatei*, *Brocchinia acuminata*, *Guzmania squarrosa*, *Stegolepis* cf. *angustata*, *Clusia* sp., *Didymiandrum stellatum*, *Racinaea spiculosa*, and others. Carnivorous plant species found in the cloud forest included *Utricularia* cf. *alpina* (Fig. 8D) and *Utricularia jamesoniana* (Fig. 8C) growing epiphytically on mossy branches. A patch of peat wetland dominated by *Stegolepis* was found and reached shortly before 8:30 AM at the elevation of approx. 1450 m a.s.l. (Fig. 7) and proved to harbor many examples of interesting endemic species typical to the Guiana Highlands, including some representatives characteristic for the Pantepui montane zone (e.g. *Heliamphora*). Most notable findings included specimens of *Orectanthe sceptrum*, *Bonnetia sessilis*, *Lindsaea stricta* var. *jamesoniiformis*, and an abundance of exceptionally large *Stegolepis ptaritepuiensis* specimens. Carnivorous plants were also common, including the following species:



Figure 7: A patch of Pantepui montane wetland dominated by *Stegolepis ptaritepuiensis* on the summit of Waukauyengtipu. 5°49'56.9"N 61°14'05.9"W, elev. ca. 1450 m a.s.l.

- *Drosera roraimae* (Fig. 8D)
- *Drosera kaieteurensis* (Fig. 8E)
- *Brocchinia reducta* (Fig. 8F)
- *Heliamphora heterodoxa* (Fig. 8H)

The occurrence of *Heliamphora heterodoxa* on the summit of Waukauyengtipu is a finding of a particular significance as it is one of just few confirmed habitats hosting a member of *Sarraceniacae* recorded in Guyana up until this day.

After spending about 1 hour exploring the area, the group was forced to retreat due to rapidly declining weather conditions and started heading towards Camp Misery shortly after 9:30 AM. The campsite was reached around 11:00 AM and cleaned up, then everyone continued moving eastwards to descend the mountain. Camp Ararata was reached at approx. 4:00 PM.

Thursday, January 10

Thursday was spent entirely on descending the trail back to Paruima. The Ararata campsite was cleaned and left before 10:00 AM and the group arrived in Paruima at approx. 2:15 PM.

Friday, January 11

On Friday, a briefing was held with the members of the Community Council of Paruima in order to share the results of the expedition. The group left Paruima by boat at 9:30 AM, accompanied by Lee Williams and Lennox Percy, as well as a boat operator. After stopping twice to see waterfalls going down the Kamarang River (Iobaru Falls – 5°49'58.5"N 61°00'51.6"W and Apurwayak Falls – 5°50'38.2"N 60°59'38.6"W) and a brief pause in Waramadong, the team reached Kamarang before 4:30 PM.

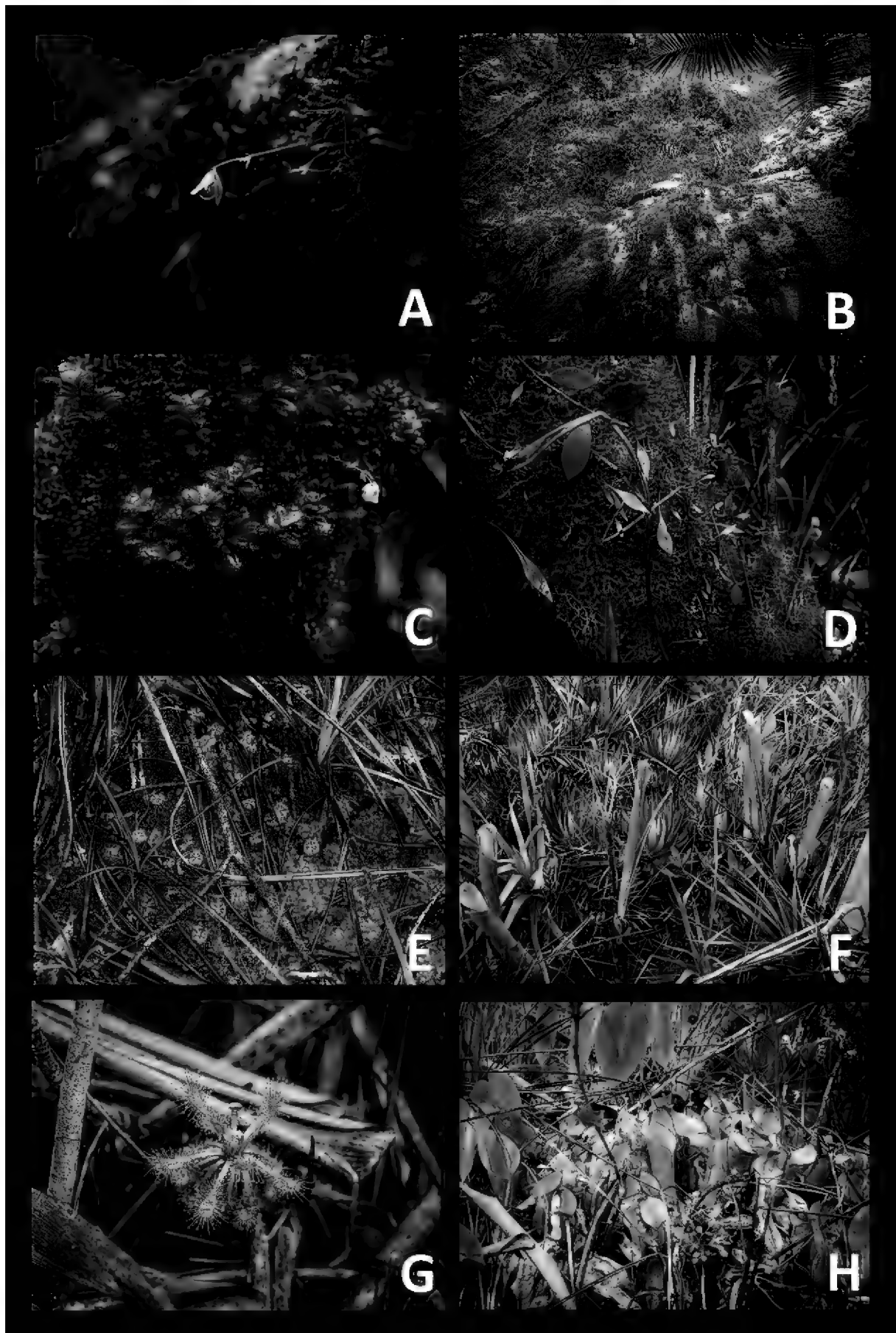


Figure 8: Carnivorous plants observed during the trek to Waukauyengtipu summit (observation perimeter No. 3, No. 4, and No. 5). A) *Utricularia jamesoniana* – found near Ararata Camp; B) Wall of Waukauyengtipu covered by vegetation; C) *Utricularia jamesoniana* on the summit of Waukauyengtipu, growing among a clump of *Macrocentrum minus*; D) *Utricularia cf. alpina*; E) *Drosera kaieteurensis*; F) *Brocchinia reducta* on the summit peat wetland, growing with *Orectanthe sceptrum*; G) *Drosera roraimae*; H) *Heliamphora heterodoxa*.

Saturday, January 12

The expedition was concluded with a return flight from Kamarang to Georgetown in the morning.

Summary of carnivorous plant observations recorded during the expedition

In total, 5 perimeters were documented. 15 species of carnivorous plants, belonging to 4 different families, were observed (Table 1).

Conclusions

Paruima and its vicinities proved to be a promising ground for future scientific and eco-tourism activities. Thanks to the high abundance of unique species of flora, the area is characterized by extraordinary opportunities for scientific research and educational projects. As for the plateau of Waukauyengtipu, although it may be a relatively small geographical feature that is overshadowed by much larger and more famous mountains in the region (e.g., Mount Roraima), it is no less interesting, not least, since it still remains very little known to this day, especially in terms of the biodiversity that it harbors. Further botanical surveying and other research undertakings are required to fully understand the biodiversity present in the vicinity of Paruima.

Acknowledgements: Members of the Pemón Arekuna community of Paruima, especially Calio Elliman, Chris Elliman, Alex Smith, Lee Williams, and Lennox Percy, as well as others who contributed to the project, are thanked for their hospitality, assistance and guidance during the January 2019 expedition. Fernando Rivadavia is thanked for his help provided with the taxonomic identification of specimens observed during the expedition, especially members of the genus *Utricularia*.

References

- Anderson, E.W. 2003. International Boundaries: A Geopolitical Atlas. New York: Routledge. pp. 356; 879.
- Hollowell, T., McDowell, T., Funk, V.A., Kelloff, C.L., and Gopaul, D. 2004. Smithsonian Plant Collections, Guyana: 1990 - 1991, Tim McDowell. Smithsonian Institution, Contributions from the United States National Herbarium; 50, pp. 24-30. <http://www.jstor.org/stable/23493110>
- Holst, B. 1997. Bromeliaceae. In: Steyermark, J.A., Berry, P.E., Holst, B.K., editors. Flora of the Venezuelan Guayana. Volume 3. Araliaceae-Cactaceae. St. Louis, Missouri Botanical Garden: Timber Press.
- Huber, O. 1995. Geography and physical features. In: Steyermark, J.A., Berry, P.E., Holst, B.K., editors. Flora of the Venezuelan Guayana. Volume 1. Introduction. St. Louis, Missouri Botanical Garden: Timber Press. pp. 36-37; 60-61.
- Kelloff, C.L., Alexander, S.N., Funk, V.A., and Clarke, H.D. 2011. Smithsonian Plant Collections, Guyana: 1995-2004, H. David Clarke. Smithsonian Contributions to Botany 97. <https://doi.org/10.5479/si.0081024X.97.1>
- McPherson, S., Wistuba, A., Fleischmann, A., and Nerz, J. 2011. Sarraceniaceae of South America. Redfern Natural History Productions Ltd.
- Robinson, A., Gibson, R., Gonella, P., McPherson, S., Nunn, R., and Fleischmann, A. 2017. *Drosera* of the World, Volume 3: Latin America & Africa. Redfern Natural History Productions Ltd.
- Taylor, P. 1989. The Genus *Utricularia*: A Taxonomic Monograph. London: HMSO, Royal Botanic Gardens, Kew.

NEW CULTIVARS

Keywords: cultivar, *Nepenthes* ‘Red Velvet Aurea’, *Nepenthes* ‘Khi Seechumpu’, *Sarracenia* ‘Bled Velvet’, *Sarracenia* ‘Pink Eye’, *Utricularia* ‘Devil Spawn’, *Utricularia* ‘King Ghidorah’.

Abstract: Six new carnivorous plant cultivars are named and described: *Nepenthes* ‘Red Velvet Aurea’, *Nepenthes* ‘Khi Seechumpu’, *Sarracenia* ‘Bled Velvet’, *Sarracenia* ‘Pink Eye’, *Utricularia* ‘Devil Spawn’, *Utricularia* ‘King Ghidorah’.

Nepenthes ‘Red Velvet Aurea’

Submitted: 20 April 2021

The seed parent of *Nepenthes* ‘Red Velvet Aurea’ is from a green clone of *Nepenthes ampullaria* originally from Thailand (Fig. 1). The pitcher of *Nepenthes* ‘Red Velvet Aurea’ is red with a velvet-like texture and a perfect roundish-oval shape about 55 × 55 mm. The spur and wing are red and feather-like with 2 to 3 lobes. The peristome is initially white and becomes bright yellow and eventually a light yellowish green with pink stripes. The pitcher mouth is oval about 35-40 mm. The neck and lid are red and shaped like an olive leaf. The stem and leaf are initially bright green, but patches of maroon appear when older. The tendril is compact.

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Figure 1: Seed parent is a green Thai clone of *Nepenthes ampullaria* (left); *Nepenthes* ‘Red Velvet Aurea’ pitcher (center) and plant (right).

Submitted: 19 May 2021

Quite some time ago I kindly received a plant of *Nepenthes orbiculata*? × *ampullaria* ‘Black Miracle’ from Mr. Alejandro Faus Payá (Bellreguard, Spain), who grew a big number of stunning hybrids of lowland tropical pitcher plants from a batch of seeds he got from Thailand. The specimen I received showed immediately some very interesting characteristics that drove me to the decision of describing the plant as a new cultivar.

The whole plant of *Nepenthes* ‘Khi Seechumpu’ is covered in a fine, velvet-like indumentum, whereas the leaves have an attractive deep red colouration abundantly blotched with maroon spots (Fig. 2). The tendril is long and thin, ending in an extremely rounded, egg-shaped pitcher from orbicular (lowers) to slightly ovate (uppers), rarely exceeding 8 cm in length. The whole trap is delightfully coloured in a deep maroon, consistent even on the wings, while the peristome is variably striped with shades of an intense burgundy. The lid is small, vertical; and is covered with faint blotches of green in a specular fashion to that found of the lamina. It appears to be a very fast and easy grower, provided that bright light, high humidity, and temperatures above 20°C are maintained.

It should be noted that while we can be sure about the male parent (*N. ampullaria* ‘Black Miracle’), the female parent’s real identity remains unknown. “Viking” is an unestablished name profusely given in Thailand to both *N. mirabilis* var. *globosa* and *N. orbiculata* and to their hybrids. I do suspect though that the latter is involved, considering the homogeneous roundness of the traps.

The name refers to a kind of Thai food called “ไข่มพู่” (usually transliterated as “*khǐsī̌chm̌phū*”) literally “red (or pink) eggs”, referring to the colour and shape of the pitchers. In order to make it easier to read, I adapted it as *N.* ‘Khi Seechumpu’, which should be pronounced in English (using IPA phonetic notation) as /kaǐˌsǐt͡ɕʰomˈpuː/.

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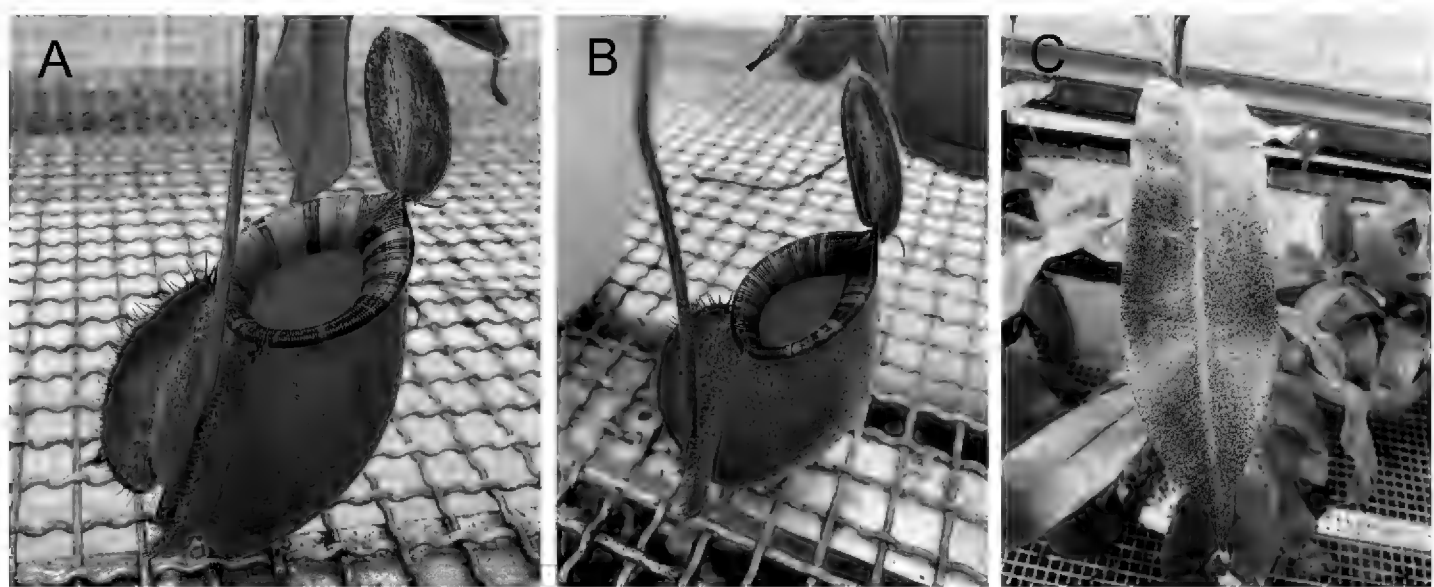


Figure 2: *Nepenthes* ‘Khi Seechumpu’ (A) lower pitcher, (B) upper pitcher, (C) leaf.

Utricularia ‘Devil Spawn’

Submitted: 11 May 2021

Utricularia ‘Devil Spawn’ (Fig. 3) is a small floriferous form of *U. bisquamata*. *Utricularia* ‘Devil Spawn’ is cultivated worldwide and is possibly the most frustrating plant in cultivation due to its ability to spread aggressively through a plant collection, hence the name coined in January 2019! It entered cultivation as *U. capensis* in the early to mid-1900s and was known as such until Taylor (1989) clarified the taxonomy on the species, including it as a synonym of the variable *U. bisquamata*. The plant spreads via threadlike subterranean rhizomes. Flower scapes are small, 1.5-2.5 cm with 1-5 small tricoloured flowers produced predominantly in late spring but flowers can often be seen year-round. The distinctive lilac, mauve, and yellow flowers create an impressive display despite their diminutive size. It is best cultivated in a peat-based mix that is kept damp to saturated year-round. *Utricularia* ‘Devil Spawn’ should be cultivated with caution and growers should consider proximity to natural wetland systems to avoid unwanted escape. Local biosecurity lists should be consulted prior to obtaining this cultivar.

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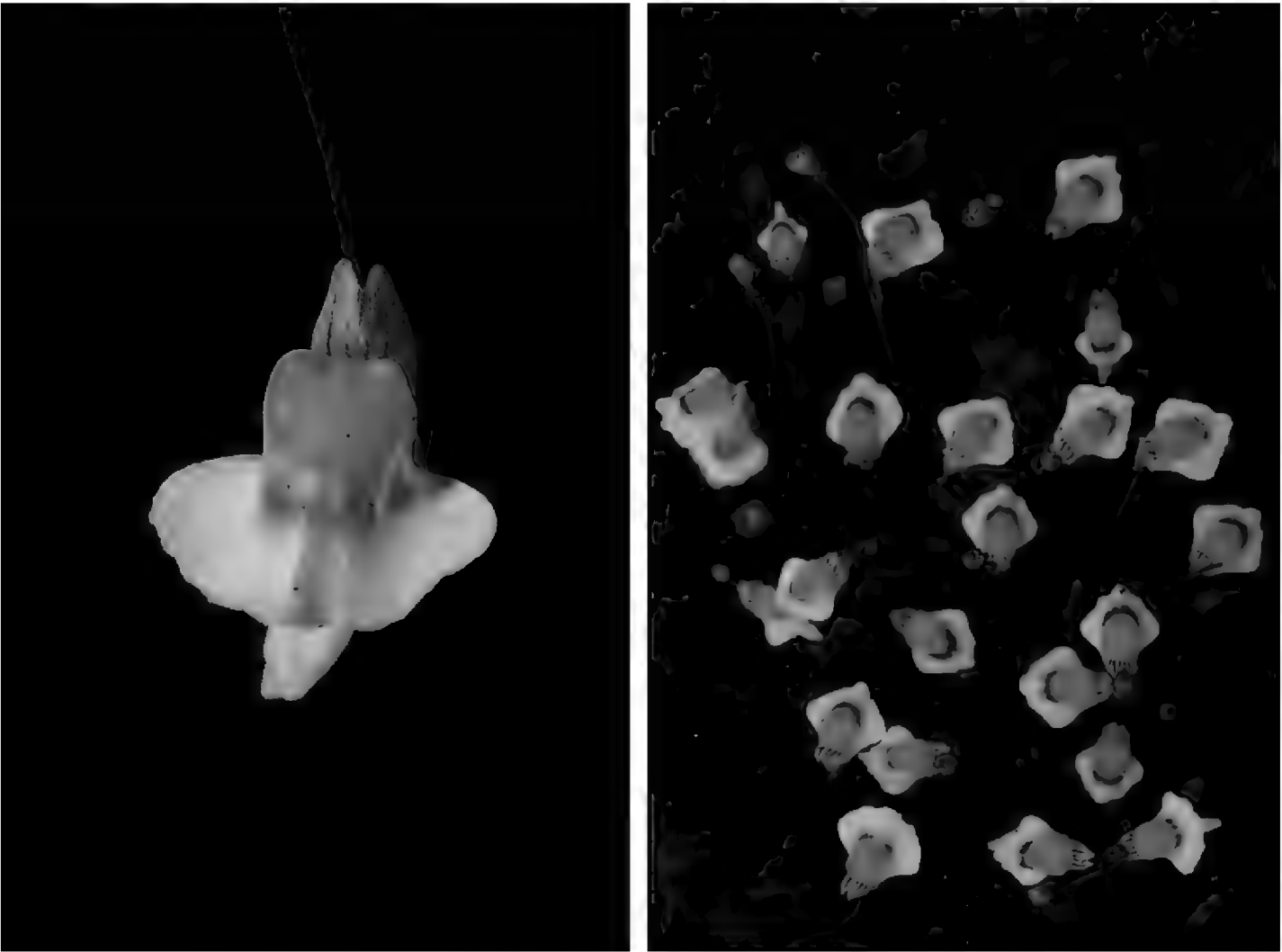


Figure 3: *Utricularia* ‘Devil Spawn’.

Utricularia ‘King Ghidorah’

Submitted: 11 May 2021

Utricularia ‘King Ghidorah’ is the result of a F2 hybrid between *U. reniformis* and *U. nelumbifolia* and selected from a large number of seedlings. It is unique in that it consistently produces flowers with three purple-edged yellow and orange central stripes at the base of the swollen lower lip and a dark purple patch in the center of the upper lip (Fig. 4). Plant is typically 10-30 cm with large reniform leaves. Flower scapes to 50 cm tall are produced from late spring to autumn with 5-12 flowers to 4 cm across. Plants spread via advantageous stolons somewhat intermediate between the two parent species in that they are neither subterranean nor rise well above the plant. Instead, the stolons rise a little above the plant before descending into the media below. This trait makes *U.* ‘King Ghidorah’ amenable to cultivation in pot culture as well as low growing water filled bromeliads. The name *U.* ‘King Ghidorah’ coined in December 2018 honours the great mythical three-headed monster that first came to earth in 1964 and has been a popular rival of Godzilla ever since.

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Figure 4: *Utricularia* ‘King Ghidorah’.

Submitted: 11 May 2021

Sarracenia ‘Bled Velvet’ is a selected seedling from the collection of Greg Bourke. It is a cross between *Sarracenia flava* from Wewahitchka area, Gulf County, Florida, USA and *S. leucophylla*. This plant exhibits fairly typical morphology for the hybrid, but it produces strikingly intense red colouration and prominent ruffling of the lid margin (Fig. 5). New pitchers are pink in colouration, transitioning to green about 1/5th of the way down the pitcher tube. Pronounced green trending to burgundy venation is prominent in the upper parts. The pitcher tube and venation ages to a deep red to almost black but the upper surface of the lid tends to lighten over time.

The name *Sarracenia* ‘Bled Velvet’ reflects the plant’s colouration and prominent venation and was coined in May 2020 by Jimmy Turner following a discussion on social media over the plant’s cultivar potential. Plants must be propagated by division.

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Figure 5: *Sarracenia* ‘Bled Velvet’ pitchers.

Sarracenia 'Pink Eye'

Submitted: 11 May 2021

Sarracenia 'Pink Eye' (Fig. 6) is a select form of the natural hybrid *Sarracenia* \times *excellens*. It was grown and sold by Australian carnivorous plant enthusiast, Phillipe Reyter throughout the late 1900's and is now relatively common in cultivation in Australia. The plant is a particularly striking clone of the hybrid in that it produces light pink pitchers with white to pale pink fenestrations on the upper rear of the pitcher body and lid. The pitchers age to dark pink/burgundy over time and the fenestrations age to dark pink, hence the name 'Pink Eye' was coined in February 2020. Plants must be propagated by division.

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Figure 6: *Sarracenia* 'Pink Eye' pitchers.



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A NEW TAXONOMIC RANK FOR *DROSERA PEDICELLARIS* (DROSERACEAE)

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For consistency with a classification using infraspecific ranks (Schlauer, J. in Carniv. Pl. Newslett. 25: 72-75, 1996), the geographically overlapping and closely related taxa *D. parvula* (syn. *D. minutiflora* sensu auct. non Planch.: Lowrie, A. in Carnivorous Plants of Australia: Magnum Opus. Redfern Natural History Productions, Poole, p. 620, 2014) and *D. pedicellaris* are combined as varieties under the older species name. A new combination is thus proposed here.

Drosera parvula var. *pedicellaris* (Lowrie) Schlauer, comb. et stat. nov.

Basionym: *Drosera pedicellaris* Lowrie, Nuytsia 15: 59 (2002).

NAMES OF CARNIVOROUS PLANT CULTIVARS REGISTERED IN 2020

Name	Publication	Date of registration
<i>Heliamphora</i> ‘Cyclops’ A.Smith	Carniv. Pl. Newslett. 49: 141 (2020)	23 Sep
<i>H.</i> ‘Scylla’ I.Bogdanow	Carniv. Pl. Newslett. 49: 93 (2020)	19 Jun
<i>Sarracenia</i> ‘Arthur Wheeler’ G.Wheeler	Pl. Carniv. 40(2): 15 (2019)	24 Jan
<i>S.</i> ‘Atlas 5’ M.Srba & M.King	Carniv. Pl. Newslett. 49: 91 (2020)	19 Jun
<i>S.</i> ‘Christophe Maerten’ L.Taerwe	Carniv. Pl. Newslett. 49: 92 (2020)	19 Jun
<i>S.</i> ‘Glynis Wheeler’ G.Wheeler	Pl. Carniv. 40(2): 11 (2019)	24 Jan
<i>S.</i> ‘Sarramphor’ A.Fisch	Carniv. Pl. Newslett. 49: 49 (2020)	21 May
<i>S.</i> ‘Super-duper’ R.Ziemer	Carniv. Pl. Newslett. 49: 140 (2020)	23 Sep
<i>S.</i> ‘Wizzleberry Toad’ J.Conner	Carniv. Pl. Newslett. 49: 50 (2020)	21 May
<i>S.</i> ‘Yvaine Little’ S.Little	Pl. Carniv. 40(2): 7 (2019)	24 Jan
<i>Cephalotus</i> ‘OG Black’ B.Lipinski	Carniv. Pl. Newslett. 49: 87 (2020)	19 Jun
<i>Dionaea</i> ‘Axe’ P.Mattevi	Carniv. Pl. Newslett. 48: 191 (2019)	24 Jan
<i>D.</i> ‘CCCP Tasmanian Devil’ C.Heath & S.Wang	Carniv. Pl. Newslett. 49: 89 (2020)	19 Jun
<i>D.</i> ‘FFT Stegosaurus’ J.Moscoso	Carniv. Pl. Newslett. 48: 188 (2019)	24 Jan
<i>D.</i> ‘Genepine’ J.Mueller	Carniv. Pl. Newslett. 48: 189 (2019)	24 Jan
<i>D.</i> ‘Morano’ P.Mattevi	Carniv. Pl. Newslett. 48: 190 (2019)	24 Jan
<i>D.</i> ‘Stove Fire’ D.Weijie	Carniv. Pl. Newslett. 49: 142 (2020)	23 Sep
<i>Pinguicula</i> ‘Eye Spy’ S.Bunclark	Carniv. Pl. Newslett. 49: 139 (2020)	23 Sep
<i>P.</i> ‘Razzberry Blonde’ M.Rubnitz	Carniv. Pl. Newslett. 49: 90 (2020)	19 Jun

